Guideline for Disinfection and Sterilization in Healthcare Facilities, 2008

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This guideline discusses use of products by healthcare personnel in healthcare settings such as hospitals, ambulatory care and home care; the recommendations are not intended for consumer use of the products discussed.

Disinfection and Sterilization in Healthcare Facilities

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EXECUTIVE SUMMARY

The Guideline for Disinfection and Sterilization in Healthcare Facilities, 2008, presents evidence-based recommendations on the preferred methods for cleaning, disinfection and sterilization of patient-care medical devices and for cleaning and disinfecting the healthcare environment. This document supercedes the relevant sections contained in the 1985 Centers for Disease Control (CDC) Guideline for Handwashing and Environmental Control. [1] Because maximum effectiveness from disinfection and sterilization results from first cleaning and removing organic and inorganic materials, this document also reviews cleaning methods. The chemical disinfectants discussed for patient-care equipment include alcohols, glutaraldehyde, formaldehyde, hydrogen peroxide, iodophors, *ortho*-phthalaldehyde, peracetic acid, phenolics, quaternary ammonium compounds, and chlorine. The choice of disinfectant, concentration, and exposure time is based on the risk for infection associated with use of the equipment and other factors discussed in this guideline. The sterilization methods discussed include steam sterilization, ethylene oxide (ETO), hydrogen peroxide gas plasma, and liquid peracetic acid. When properly used, these cleaning, disinfection, and sterilization processes can reduce the risk for infection associated with use of invasive and noninvasive medical and surgical devices. However, for these processes to be effective, health-care workers should adhere strictly to the cleaning, disinfection, and sterilization recommendations in this document and to instructions on product labels.

In addition to updated recommendations, new topics addressed in this guideline include 1) inactivation of antibiotic-resistant bacteria, bioterrorist agents, emerging pathogens, and bloodborne pathogens; 2) toxicologic, environmental, and occupational concerns associated with disinfection and sterilization practices; 3) disinfection of patient-care equipment used in ambulatory settings and home care; 4) new sterilization processes, such as hydrogen peroxide gas plasma and liquid peracetic acid; and 5) disinfection of complex medical instruments (e.g., endoscopes).

INTRODUCTION

In the United States, approximately 46.5 million surgical procedures and even more invasive medical procedures—including approximately 5 million gastrointestinal endoscopies—are performed each year. [2] Each procedure involves contact by a medical device or surgical instrument with a patient's sterile tissue or mucous membranes. A major risk of all such procedures is the introduction of pathogens that can lead to infection. Failure to properly disinfect or sterilize equipment carries not only risk associated with breach of host barriers but also risk for person-to-person transmission (e.g., hepatitis B virus) and transmission of environmental pathogens (e.g., *Pseudomonas aeruginosa*).

Disinfection and sterilization are essential for ensuring that medical and surgical instruments do not transmit infectious pathogens to patients. Because sterilization of all patient-care items is not necessary, health-care policies must identify, primarily on the basis of the items' intended use, whether cleaning, disinfection, or sterilization is indicated.

Multiple studies in many countries have documented lack of compliance with established guidelines for disinfection and sterilization. [3-6] Failure to comply with scientifically-based guidelines has led to numerous outbreaks. [6-12] This guideline presents a pragmatic approach to the judicious selection and proper use of disinfection and sterilization processes; the approach is based on well-designed studies assessing the efficacy (through laboratory investigations) and effectiveness (through clinical studies) of disinfection and sterilization procedures.

METHODS

This guideline resulted from a review of all MEDLINE articles in English listed under the MeSH headings of *disinfection* or *sterilization* (focusing on health-care equipment and supplies) from January 1980 through August 2006. References listed in these articles also were reviewed. Selected articles published before 1980 were reviewed and, if still relevant, included in the guideline. The three major peer-reviewed journals in infection control—*American Journal of Infection Control, Infection Control and Hospital Epidemiology,* and *Journal of Hospital Infection*—were searched for relevant articles published from January 1990 through August 2006. Abstracts presented at the annual meetings of the Society for Healthcare Epidemiology of America and Association for professionals in Infection Control and Epidemiology, Inc. during 1997–2006 also were reviewed; however, abstracts were not used to support the recommendations.

DEFINITION OF TERMS

Sterilization describes a process that destroys or eliminates all forms of microbial life and is carried out in health-care facilities by physical or chemical methods. Steam under pressure, dry heat, EtO gas, hydrogen peroxide gas plasma, and liquid chemicals are the principal sterilizing agents used in health-care facilities. Sterilization is intended to convey an absolute meaning; unfortunately, however, some health professionals and the technical and commercial literature refer to "disinfection" as "sterilization" and items as "partially sterile." When chemicals are used to destroy all forms of microbiologic life, they can be called chemical sterilants. These same germicides used for shorter exposure periods also can be part of the disinfection process (i.e., high-level disinfection).

Disinfection describes a process that eliminates many or all pathogenic microorganisms, except bacterial spores, on inanimate objects (Tables 1 and 2). In health-care settings, objects usually are disinfected by liquid chemicals or wet pasteurization. Each of the various factors that affect the efficacy of

disinfection can nullify or limit the efficacy of the process.

Factors that affect the efficacy of both disinfection and sterilization include prior cleaning of the object; organic and inorganic load present; type and level of microbial contamination; concentration of and exposure time to the germicide; physical nature of the object (e.g., crevices, hinges, and lumens); presence of biofilms; temperature and pH of the disinfection process; and in some cases, relative humidity of the sterilization process (e.g., ethylene oxide).

Unlike sterilization, disinfection is not sporicidal. A few disinfectants will kill spores with prolonged exposure times (3–12 hours); these are called *chemical sterilants*. At similar concentrations but with shorter exposure periods (e.g., 20 minutes for 2% glutaraldehyde), these same disinfectants will kill all microorganisms except large numbers of bacterial spores; they are called *high-level disinfectants*. *Low-level disinfectants* can kill most vegetative bacteria, some fungi, and some viruses in a practical period of time (≤10 minutes). *Intermediate-level disinfectants* might be cidal for mycobacteria, vegetative bacteria, most viruses, and most fungi but do not necessarily kill bacterial spores. Germicides differ markedly, primarily in their antimicrobial spectrum and rapidity of action.

Cleaning is the removal of visible soil (e.g., organic and inorganic material) from objects and surfaces and normally is accomplished manually or mechanically using water with detergents or enzymatic products. Thorough cleaning is essential before high-level disinfection and sterilization because inorganic and organic materials that remain on the surfaces of instruments interfere with the effectiveness of these processes. *Decontamination* removes pathogenic microorganisms from objects so they are safe to handle, use, or discard.

Terms with the suffix *cide* or *cidal* for killing action also are commonly used. For example, a germicide is an agent that can kill microorganisms, particularly pathogenic organisms (“germs”). The term *germicide* includes both antiseptics and disinfectants. *Antiseptics* are germicides applied to living tissue and skin; *disinfectants* are antimicrobials applied only to inanimate objects. In general, antiseptics are used only on the skin and not for surface disinfection, and disinfectants are not used for skin antisepsis because they can injure skin and other tissues. Virucide, fungicide, bactericide, sporicide, and tuberculocide can kill the type of microorganism identified by the prefix. For example, a bactericide is an agent that kills bacteria. [13-18]

A RATIONAL APPROACH TO DISINFECTION AND STERILIZATION

More than 30 years ago, Earle H. Spaulding devised a rational approach to disinfection and sterilization of patient-care items and equipment.[14] This classification scheme is so clear and logical that it has been retained, refined, and successfully used by infection control professionals and others when planning methods for disinfection or sterilization. [1, 13, 15, 17, 19, 20] Spaulding believed the nature of disinfection could be understood readily if instruments and items for patient care were categorized as critical, semicritical, and noncritical according to the degree of risk for infection involved in use of the items. The CDC *Guideline for Handwashing and Hospital Environmental Control* [21], *Guidelines for the Prevention of Transmission of Human Immunodeficiency Virus (HIV) and Hepatitis B Virus (HBV) to Health-Care and Public-Safety Workers*[22], and *Guideline for Environmental Infection Control in Health-Care Facilities*[23] employ this terminology.

Critical Items

Critical items confer a high risk for infection if they are contaminated with any microorganism. Thus, objects that enter sterile tissue or the vascular system must be sterile because any microbial contamination could transmit disease. This category includes surgical instruments, cardiac and urinary catheters, implants, and ultrasound probes used in sterile body cavities. Most of the items in this category should be purchased as sterile or be sterilized with steam if possible. Heat-sensitive objects can be treated with EtO, hydrogen peroxide gas plasma; or if other methods are unsuitable, by liquid chemical sterilants. Germicides categorized as chemical sterilants include $\geq$2.4% glutaraldehyde-based formulations, 0.95% glutaraldehyde with 1.64% phenol/phenate, 7.5% stabilized hydrogen peroxide, 7.35% hydrogen peroxide with 0.23% peracetic acid, 0.2% peracetic acid, and 0.08% peracetic acid with 1.0% hydrogen peroxide. Liquid chemical sterilants reliably produce sterility only if cleaning precedes treatment and if proper guidelines are followed regarding concentration, contact time, temperature, and pH.

Semicritical Items

Semicritical items contact mucous membranes or nonintact skin. This category includes respiratory therapy and anesthesia equipment, some endoscopes, laryngoscope blades [24], esophageal manometry probes, cystoscopes [25], anorectal manometry catheters, and diaphragm fitting rings. These medical devices should be free from all microorganisms; however, small numbers of bacterial spores are permissible. Intact mucous membranes, such as those of the lungs and the gastrointestinal tract, generally are resistant to infection by common bacterial spores but susceptible to other organisms, such as bacteria, mycobacteria, and viruses. Semicritical items minimally require high-level disinfection using chemical disinfectants. Glutaraldehyde, hydrogen peroxide, *ortho*-phthalaldehyde, and peracetic acid with hydrogen peroxide are cleared by the Food and Drug Administration (FDA) and are dependable high-level disinfectants provided the factors influencing germicidal procedures are met (Table 1). When a disinfectant is selected for use with certain patient-care items, the chemical compatibility after extended use with the items to be disinfected also must be considered.

High-level disinfection traditionally is defined as complete elimination of all microorganisms in or on an instrument, except for small numbers of bacterial spores. The FDA definition of high-level disinfection is a sterilant used for a shorter contact time to achieve a 6-$\log_{10}$ kill of an appropriate *Mycobacterium* species. Cleaning followed by high-level disinfection should eliminate enough pathogens to prevent transmission of infection. [26, 27]

Laparoscopes and arthroscopes entering sterile tissue ideally should be sterilized between patients. However, in the United States, this equipment sometimes undergoes only high-level disinfection between patients. [28-30] As with flexible endoscopes, these devices can be difficult to clean and high-level disinfect or sterilize because of intricate device design (e.g., long narrow lumens, hinges). Meticulous

cleaning must precede any high-level disinfection or sterilization process. Although sterilization is preferred, no reports have been published of outbreaks resulting from high-level disinfection of these scopes when they are properly cleaned and high-level disinfected. Newer models of these instruments can withstand steam sterilization that for critical items would be preferable to high-level disinfection.

Rinsing endoscopes and flushing channels with sterile water, filtered water, or tap water will prevent adverse effects associated with disinfectant retained in the endoscope (e.g., disinfectant-induced colitis). Items can be rinsed and flushed using sterile water after high-level disinfection to prevent contamination with organisms in tap water, such as nontuberculous mycobacteria, [10, 31, 32] *Legionella*, [33-35] or gram-negative bacilli such as *Pseudomonas*. [1, 17, 36-38] Alternatively, a tapwater or filtered water (0.2μ filter) rinse should be followed by an alcohol rinse and forced air drying. [28, 38-40] Forced-air drying markedly reduces bacterial contamination of stored endoscopes, most likely by removing the wet environment favorable for bacterial growth. [39] After rinsing, items should be dried and stored (e.g., packaged) in a manner that protects them from recontamination.

Some items that may come in contact with nonintact skin for a brief period of time (i.e., hydrotherapy tanks, bed side rails) are usually considered noncritical surfaces and are disinfected with intermediate-level disinfectants (i.e., phenolic, iodophor, alcohol, chlorine) [23]. Since hydrotherapy tanks have been associated with spread of infection, some facilities have chosen to disinfect them with recommended levels of chlorine [23, 41].

In the past, high-level disinfection was recommended for mouthpieces and spirometry tubing (e.g., glutaraldehyde) but cleaning the interior surfaces of the spirometers was considered unnecessary. [42] This was based on a study that showed that mouthpieces and spirometry tubing become contaminated with microorganisms but there was no bacterial contamination of the surfaces inside the spirometers. Filters have been used to prevent contamination of this equipment distal to the filter; such filters and the proximal mouthpiece are changed between patients.

Noncritical Items

Noncritical items are those that come in contact with intact skin but not mucous membranes. Intact skin acts as an effective barrier to most microorganisms; therefore, the sterility of items coming in contact with intact skin is "not critical." In this guideline, noncritical items are divided into noncritical patient care items and noncritical environmental surfaces [43, 44]. Examples of noncritical patient-care items are bedpans, blood pressure cuffs, crutches and computers [45]. In contrast to critical and some semicritical items, most noncritical reusable items may be decontaminated where they are used and do not need to be transported to a central processing area. Virtually no risk has been documented for transmission of infectious agents to patients through noncritical items [37] when they are used as noncritical items and do not contact non-intact skin and/or mucous membranes. Table 1 lists several low-level disinfectants that may be used for noncritical items. Most Environmental Protection Agency (EPA)-registered disinfectants have a 10-minute label claim. However, multiple investigators have demonstrated the effectiveness of these disinfectants against vegetative bacteria (e.g., *Listeria*, *Escherichia coli*, *Salmonella*, vancomycin-resistant Enterococci, methicillin-resistant *Staphylococcus aureus*), yeasts (e.g., *Candida*), mycobacteria (e.g., *Mycobacterium tuberculosis*), and viruses (e.g. poliovirus) at exposure times of 30–60 seconds[46-64] Federal law requires all applicable label instructions on EPA-registered products to be followed (e.g., use-dilution, shelf life, storage, material compatibility, safe use, and disposal). If the user selects exposure conditions (e.g., exposure time) that differ from those on the EPA-registered products label, the user assumes liability for any injuries resulting from off-label use and is potentially subject to enforcement action under Federal Insecticide, Fungicide, and Rodenticide Act (FIFRA) [65].

Noncritcal environmental surfaces include bed rails, some food utensils, bedside tables, patient furniture and floors. Noncritical environmental surfaces frequently touched by hand (e.g., bedside tables,

bed rails) potentially could contribute to secondary transmission by contaminating hands of health-care workers or by contacting medical equipment that subsequently contacts patients [13, 46-48, 51, 66, 67]. Mops and reusable cleaning cloths are regularly used to achieve low-level disinfection on environmental surfaces. However, they often are not adequately cleaned and disinfected, and if the water-disinfectant mixture is not changed regularly (e.g., after every three to four rooms, at no longer than 60-minute intervals), the mopping procedure actually can spread heavy microbial contamination throughout the health-care facility [68]. In one study, standard laundering provided acceptable decontamination of heavily contaminated mopheads but chemical disinfection with a phenolic was less effective. [68] Frequent laundering of mops (e.g., daily), therefore, is recommended. Single-use disposable towels impregnated with a disinfectant also can be used for low-level disinfection when spot-cleaning of noncritical surfaces is needed[45].

Changes in Disinfection and Sterilization Since 1981

The Table in the CDC *Guideline for Environmental Control* prepared in 1981 as a guide to the appropriate selection and use of disinfectants has undergone several important changes (Table 1). [15] First, formaldehyde-alcohol has been deleted as a recommended chemical sterilant or high-level disinfectant because it is irritating and toxic and not commonly used. Second, several new chemical sterilants have been added, including hydrogen peroxide, peracetic acid [58, 69, 70], and peracetic acid and hydrogen peroxide in combination. Third, 3% phenolics and iodophors have been deleted as high-level disinfectants because of their unproven efficacy against bacterial spores, *M. tuberculosis,* and/or some fungi. [55, 71] Fourth, isopropyl alcohol and ethyl alcohol have been excluded as high-level disinfectants [15] because of their inability to inactivate bacterial spores and because of the inability of isopropyl alcohol to inactivate hydrophilic viruses (i.e., poliovirus, coxsackie virus). [72] Fifth, a 1:16 dilution of 2.0% glutaraldehyde-7.05% phenol-1.20% sodium phenate (which contained 0.125% glutaraldehyde, 0.440% phenol, and 0.075% sodium phenate when diluted) has been deleted as a high-level disinfectant because this product was removed from the marketplace in December 1991 because of a lack of bactericidal activity in the presence of organic matter; a lack of fungicidal, tuberculocidal and sporicidal activity; and reduced virucidal activity. [49, 55, 56, 71, 73-79] Sixth, the exposure time required to achieve high-level disinfection has been changed from 10-30 minutes to 12 minutes or more depending on the FDA-cleared label claim and the scientific literature. [27, 55, 69, 76, 80-84] A glutaraldehyde and an ortho-phthalaldehyde have an FDA-cleared label claim of 5 minutes when used at 35°C and 25°C, respectively, in an automated endoscope reprocessor with FDA-cleared capability to maintain the solution at the appropriate temperature. [85]

In addition, many new subjects have been added to the guideline. These include inactivation of emerging pathogens, bioterrorist agents, and bloodborne pathogens; toxicologic, environmental, and occupational concerns associated with disinfection and sterilization practices; disinfection of patient-care equipment used in ambulatory and home care; inactivation of antibiotic-resistant bacteria; new sterilization processes, such as hydrogen peroxide gas plasma and liquid peracetic acid; and disinfection of complex medical instruments (e.g., endoscopes).

DISINFECTION OF HEALTHCARE EQUIPMENT

Concerns about Implementing the Spaulding Scheme

One problem with implementing the aforementioned scheme is oversimplification. For example, the scheme does not consider problems with reprocessing of complicated medical equipment that often is heat-sensitive or problems of inactivating certain types of infectious agents (e.g., prions, such as Creutzfeldt-Jakob disease [CJD] agent). Thus, in some situations, choosing a method of disinfection remains difficult, even after consideration of the categories of risk to patients. This is true particularly for a few medical devices (e.g., arthroscopes, laparoscopes) in the critical category because of controversy about whether they should be sterilized or high-level disinfected. [28, 86] Heat-stable scopes (e.g., many rigid scopes) should be steam sterilized. Some of these items cannot be steam sterilized because they are heat-sensitive; additionally, sterilization using ethylene oxide (EtO) can be too time-consuming for routine use between patients (new technologies, such as hydrogen peroxide gas plasma and peracetic acid reprocessor, provide faster cycle times). However, evidence that sterilization of these items improves patient care by reducing the infection risk is lacking[29, 87-91]. Many newer models of these instruments can withstand steam sterilization, which for critical items is the preferred method.

Another problem with implementing the Spaulding scheme is processing of an instrument in the semicritical category (e.g., endoscope) that would be used in conjunction with a critical instrument that contacts sterile body tissues. For example, is an endoscope used for upper gastrointestinal tract investigation still a semicritical item when used with sterile biopsy forceps or in a patient who is bleeding heavily from esophageal varices? Provided that high-level disinfection is achieved, and all microorganisms except bacterial spores have been removed from the endoscope, the device should not represent an infection risk and should remain in the semicritical category [92-94]. Infection with spore-forming bacteria has not been reported from appropriately high-level disinfected endoscopes.

An additional problem with implementation of the Spaulding system is that the optimal contact time for high-level disinfection has not been defined or varies among professional organizations, resulting in different strategies for disinfecting different types of semicritical items (e.g., endoscopes, applanation tonometers, endocavitary transducers, cryosurgical instruments, and diaphragm fitting rings). Until simpler and effective alternatives are identified for device disinfection in clinical settings, following this guideline, other CDC guidelines [1, 22, 95, 96] and FDA-cleared instructions for the liquid chemical sterilants/high-level disinfectants would be prudent.

Reprocessing of Endoscopes

Physicians use endoscopes to diagnose and treat numerous medical disorders. Even though endoscopes represent a valuable diagnostic and therapeutic tool in modern medicine and the incidence of infection associated with their use reportedly is very low (about 1 in 1.8 million procedures) [97], more healthcare–associated outbreaks have been linked to contaminated endoscopes than to any other medical device [6-8, 12, 98]. To prevent the spread of health-care–associated infections, all heat-sensitive endoscopes (e.g., gastrointestinal endoscopes, bronchoscopes, nasopharygoscopes) must be properly cleaned and, at a minimum, subjected to high-level disinfection after each use. High-level disinfection can be expected to destroy all microorganisms, although when high numbers of bacterial spores are present, a few spores might survive.

Because of the types of body cavities they enter, flexible endoscopes acquire high levels of microbial contamination (bioburden) during each use [99]. For example, the bioburden found on flexible gastrointestinal endoscopes after use has ranged from 10^5 colony forming units (CFU)/mL to 10^{10} CFU/mL, with the highest levels found in the suction channels [99-102]. The average load on bronchoscopes before cleaning was 6.4×10^4 CFU/mL. Cleaning reduces the level of microbial contamination by 4–6 $\log_{10}$ [83, 103]. Using human immunovirus (HIV)-contaminated endoscopes, several investigators have shown that cleaning completely eliminates the microbial contamination on the scopes [104, 105]. Similarly, other investigators found that EtO sterilization or soaking in 2% glutaraldehyde for 20 minutes was effective only when the device first was properly cleaned [106].

FDA maintains a list of cleared liquid chemical sterilants and high-level disinfectants that can be used to reprocess heat-sensitive medical devices, such as flexible endoscopes (http://www.fda.gov/cdrh/ode/germlab.html). At this time, the FDA-cleared and marketed formulations include: ≥2.4% glutaraldehyde, 0.55% *ortho*-phthalaldehyde (OPA), 0.95% glutaraldehyde with 1.64% phenol/phenate, 7.35% hydrogen peroxide with 0.23% peracetic acid, 1.0% hydrogen peroxide with 0.08% peracetic acid, and 7.5% hydrogen peroxide [85]. These products have excellent antimicrobial activity; however, some oxidizing chemicals (e.g., 7.5% hydrogen peroxide, and 1.0% hydrogen peroxide with 0.08% peracetic acid [latter product is no longer marketed]) reportedly have caused cosmetic and functional damage to endoscopes [69]. Users should check with device manufacturers for information about germicide compatibility with their device. If the germicide is FDA-cleared, then it is safe when used according to label directions; however, professionals should review the scientific literature for newly available data regarding human safety or materials compatibility. EtO sterilization of flexible endoscopes is infrequent because it requires a lengthy processing and aeration time (e.g., 12 hours) and is a potential hazard to staff and patients. The two products most commonly used for reprocessing endoscopes in the United States are glutaraldehyde and an automated, liquid chemical sterilization process that uses peracetic acid [107]. The American Society for Gastrointestinal Endoscopy (ASGE) recommends glutaraldehyde solutions that do not contain surfactants because the soapy residues of surfactants are difficult to remove during rinsing [108]. *ortho*-phthalaldehyde has begun to replace glutaraldehyde in many health-care facilities because it has several potential advantages over glutaraldehyde: is not known to irritate the eyes and nasal passages, does not require activation or exposure monitoring, and has a 12-minute high-level disinfection claim in the United States [69]. Disinfectants that are not FDA-cleared and should not be used for reprocessing endoscopes include iodophors, chlorine solutions, alcohols, quaternary ammonium compounds, and phenolics. These solutions might still be in use outside the United States, but their use should be strongly discouraged because of lack of proven efficacy against all microorganisms or materials incompatibility.

FDA clearance of the contact conditions listed on germicide labeling is based on the manufacturer's test results (http://www.fda.gov/cdrh/ode/germlab.html). Manufacturers test the product under worst-case conditions for germicide formulation (i.e., minimum recommended concentration of the active ingredient), and include organic soil. Typically manufacturers use 5% serum as the organic soil and hard water as examples of organic and inorganic challenges. The soil represents the organic loading to which the device is exposed during actual use and that would remain on the device in the absence of cleaning. This method ensures that the contact conditions completely eliminate the test mycobacteria (e.g., 10^5 to 10^6 *Mycobacteria tuberculosis* in organic soil and dried on a scope) if inoculated in the most difficult areas for the disinfectant to penetrate and contact in the absence of cleaning and thus provides a margin of safety [109]. For 2.4% glutaraldehyde that requires a 45-minute immersion at 25°C to achieve high-level disinfection (i.e., 100% kill of *M. tuberculosis*). FDA itself does not conduct testing but relies solely on the disinfectant manufacturer's data. Data suggest that *M. tuberculosis* levels can be reduced by at least 8 $\log_{10}$ with cleaning (4 $\log_{10}$) [83, 101, 102, 110], followed by chemical disinfection for 20 minutes at 20°C (4 to 6 $\log_{10}$) [83, 93, 111, 112]. On the basis of these data, APIC [113], the Society of Gastroenterology Nurses and Associates (SGNA) [38, 114, 115], the ASGE [108], American College of Chest Physicians [12], and a multi-society guideline [116] recommend alternative contact conditions with 2% glutaraldehyde to achieve high-level disinfection (e.g., that equipment be immersed in 2% glutaraldehyde at 20°C for at least 20 minutes for high-level disinfection). Federal regulations are to follow the FDA-cleared label claim for high-level disinfectants. The FDA-cleared labels for high-level disinfection with >2% glutaraldehyde at 25°C range from 20-90 minutes, depending upon the product based on three tier testing which includes AOAC sporicidal tests, simulated use testing with mycobacterial and in-use testing. The studies supporting the efficacy of >2% glutaraldehyde for 20 minutes at 20ºC assume adequate cleaning prior to disinfection, whereas the FDA-cleared label claim incorporates an added margin of safety to accommodate possible lapses in cleaning practices. Facilities that have chosen to apply the 20 minute duration at 20ºC have done so based on the IA recommendation in the July 2003 SHEA position paper, "Multi-society Guideline for Reprocessing Flexible Gastrointestinal Endoscopes" [19, 57, 83, 94, 108, 111, 116-121].

Flexible endoscopes are particularly difficult to disinfect [122] and easy to damage because of their intricate design and delicate materials. [123] Meticulous cleaning must precede any sterilization or high-level disinfection of these instruments. Failure to perform good cleaning can result in sterilization or disinfection failure, and outbreaks of infection can occur. Several studies have demonstrated the importance of cleaning in experimental studies with the duck hepatitis B virus (HBV) [106, 124], HIV [125]and *Helicobacter pylori*. [126]

An examination of health-care–associated infections related only to endoscopes through July 1992 found 281 infections transmitted by gastrointestinal endoscopy and 96 transmitted by bronchoscopy. The clinical spectrum ranged from asymptomatic colonization to death. *Salmonella* species and *Pseudomonas aeruginosa* repeatedly were identified as causative agents of infections transmitted by gastrointestinal endoscopy, and *M. tuberculosis*, atypical mycobacteria, and *P. aeruginosa* were the most common causes of infections transmitted by bronchoscopy [12]. Major reasons for transmission were inadequate cleaning, improper selection of a disinfecting agent, and failure to follow recommended cleaning and disinfection procedures [6, 8, 37, 98], and flaws in endoscope design [127, 128] or automated endoscope reprocessors. [7, 98] Failure to follow established guidelines has continued to result in infections associated with gastrointestinal endoscopes [8] and bronchoscopes [7, 12]. Potential device-associated problems should be reported to the FDA Center for Devices and Radiologic Health. One multistate investigation found that 23.9% of the bacterial cultures from the internal channels of 71 gastrointestinal endoscopes grew ≥100,000 colonies of bacteria after completion of all disinfection and sterilization procedures (nine of 25 facilities were using a product that has been removed from the marketplace [six facilities using 1:16 glutaraldehyde phenate], is not FDA-cleared as a high-level disinfectant [an iodophor] or no disinfecting agent) and before use on the next patient[129]. The incidence of postendoscopic procedure infections from an improperly processed endoscope has not been rigorously assessed.

Automated endoscope reprocessors (AER) offer several advantages over manual reprocessing: they automate and standardize several important reprocessing steps[130-132], reduce the likelihood that an essential reprocessing step will be skipped, and reduce personnel exposure to high-level disinfectants or chemical sterilants. Failure of AERs has been linked to outbreaks of infections [133] or colonization [7, 134], and the AER water filtration system might not be able to reliably provide “sterile” or bacteria-free rinse water[135, 136]. Establishment of correct connectors between the AER and the device is critical to ensure complete flow of disinfectants and rinse water [7, 137]. In addition, some endoscopes such as the duodenoscopes (e.g., endoscopic retrograde cholangiopancreatography [ERCP]) contain features (e.g., elevator-wire channel) that require a flushing pressure that is not achieved by most AERs and must be reprocessed manually using a 2- to 5-mL syringe, until new duodenoscopes equipped with a wider elevator-channel that AERs can reliably reprocess become available [132]. Outbreaks involving removable endoscope parts [138, 139] such as suction valves and endoscopic accessories designed to be inserted through flexible endoscopes such as biopsy forceps emphasize the importance of cleaning to remove all foreign matter before high-level disinfection or sterilization. [140] Some types of valves are now available as single-use, disposable products (e.g., bronchoscope valves) or steam sterilizable products (e.g., gastrointestinal endoscope valves).

AERs need further development and redesign [7, 141], as do endoscopes [123, 142], so that they do not represent a potential source of infectious agents. Endoscopes employing disposable components (e.g., protective barrier devices or sheaths) might provide an alternative to conventional liquid chemical high-level disinfection/sterilization[143, 144]. Another new technology is a swallowable camera-in-a-capsule that travels through the digestive tract and transmits color pictures of the small intestine to a receiver worn outside the body. This capsule currently does not replace colonoscopies**.**

Published recommendations for cleaning and disinfecting endoscopic equipment should be strictly followed [12, 38, 108, 113-116, 145-148]. Unfortunately, audits have shown that personnel do not consistently adhere to guidelines on reprocessing [149-151] and outbreaks of infection continue to occur. [152-154] To ensure

reprocessing personnel are properly trained, each person who reprocesses endoscopic instruments should receive initial and annual competency testing [38, 155].

In general, endoscope disinfection or sterilization with a liquid chemical sterilant involves five steps after leak testing:

1. Clean: mechanically clean internal and external surfaces, including brushing internal channels and flushing each internal channel with water and a detergent or enzymatic cleaners (leak testing is recommended for endoscopes before immersion).
2. Disinfect: immerse endoscope in high-level disinfectant (or chemical sterilant) and perfuse (eliminates air pockets and ensures contact of the germicide with the internal channels) disinfectant into all accessible channels, such as the suction/biopsy channel and air/water channel and expose for a time recommended for specific products.
3. Rinse: rinse the endoscope and all channels with sterile water, filtered water (commonly used with AERs) or tap water (i.e., high-quality potable water that meets federal clean water standards at the point of use).
4. Dry: rinse the insertion tube and inner channels with alcohol, and dry with forced air after disinfection and before storage.

Store: store the endoscope in a way that prevents recontamination and promotes drying (e.g., hung vertically). Drying the endoscope (steps 3 and 4) is essential to greatly reduce the chance of recontamination of the endoscope by microorganisms that can be present in the rinse water [116, 156]. One study demonstrated that reprocessed endoscopes (i.e., air/water channel, suction/biopsy channel) generally were negative (100% after 24 hours; 90% after 7 days [1 CFU of coagulase-negative *Staphylococcus* in one channel]) for bacterial growth when stored by hanging vertically in a ventilated cabinet[157]. Other investigators found all endoscopes were bacteria-free immediately after high-level disinfection, and only four of 135 scopes were positive during the subsequent 5-day assessment (skin bacteria cultured from endoscope surfaces). All flush-through samples remained sterile [158]. Because tapwater can contain low levels of microorganisms[159], some researchers have suggested that only sterile water (which can be prohibitively expensive) [160] or AER filtered water be used. The suggestion to use only sterile water or filtered water is not consistent with published guidelines that allow tapwater with an alcohol rinse and forced air-drying [38, 108, 113] or the scientific literature. [39, 93] In addition, no evidence of disease transmission has been found when a tap water rinse is followed by an alcohol rinse and forced-air drying. AERs produce filtered water by passage through a bacterial filter (e.g., 0.2 μ). Filtered rinse water was identified as a source of bacterial contamination in a study that cultured the accessory and suction channels of endoscopes and the internal chambers of AERs during 1996–2001 and reported 8.7% of samples collected during 1996–1998 had bacterial growth, with 54% being *Pseudomonas* species. After a system of hot water flushing of the piping (60°C for 60 minutes daily) was introduced, the frequency of positive cultures fell to approximately 2% with only rare isolation of >10 CFU/mL [161]. In addition to the endoscope reprocessing steps, a protocol should be developed that ensures the user knows whether an endoscope has been appropriately cleaned and disinfected (e.g., using a room or cabinet for processed endoscopes only) or has not been reprocessed. When users leave endoscopes on movable carts, confusion can result about whether the endoscope has been processed. Although one guideline recommended endoscopes (e.g., duodenoscopes) be reprocessed immediately before use [147], other guidelines do not require this activity [38, 108, 115] and except for the Association of periOperative Registered Nurses (AORN), professional organizations do not recommended that reprocessing be repeated as long as the original processing is done correctly. As part of a quality assurance program, healthcare facility personnel can consider random bacterial surveillance cultures of processed endoscopes to ensure high-level disinfection or sterilization[7, 162-164]. Reprocessed endoscopes should be free of microbial pathogens except for small numbers of relatively avirulent microbes that represent exogenous environmental contamination (e.g., coagulase-negative *Staphylococcus*, *Bacillus* species, diphtheroids). Although recommendations exist for the final rinse water used during endoscope reprocessing to be microbiologically cultured at least monthly [165], a microbiologic standard has not been

set, and the value of routine endoscope cultures has not been shown [166]. In addition, neither the routine culture of reprocessed endoscopes nor the final rinse water has been validated by correlating viable counts on an endoscope to infection after an endoscopic procedure. If reprocessed endoscopes were cultured, sampling the endoscope would assess water quality and other important steps (e.g., disinfectant effectiveness, exposure time, cleaning) in the reprocessing procedure. A number of methods for sampling endoscopes and water have been described [23, 157, 161, 163, 167, 168]. Novel approaches (e.g., detection of adenosine triphosphate [ATP]) to evaluate the effectiveness of endoscope cleaning [169, 170] or endoscope reprocessing [171] also have been evaluated, but no method has been established as a standard for assessing the outcome of endoscope reprocessing.

The carrying case used to transport clean and reprocessed endoscopes outside the health-care environment should not be used to store an endoscope or to transport the instrument within the health-care facility. A contaminated endoscope should never be placed in the carrying case because the case can also become contaminated. When the endoscope is removed from the case, properly reprocessed, and put back in the case, the case could recontaminate the endoscope. A contaminated carrying case should be discarded (Olympus America, June 2002, written communication).

Infection-control professionals should ensure that institutional policies are consistent with national guidelines and conduct infection-control rounds periodically (e.g., at least annually) in areas where endoscopes are reprocessed to ensure policy compliance. Breaches in policy should be documented and corrective action instituted. In incidents in which endoscopes were not exposed to a high-level disinfection process, patients exposed to potentially contaminated endoscopes have been assessed for possible acquisition of HIV, HBV, and hepatitis C virus (HCV). A 14-step method for managing a failure incident associated with high-level disinfection or sterilization has been described [Rutala WA, 2006 #12512]. The possible transmission of bloodborne and other infectious agents highlights the importance of rigorous infection control[172, 173].

Laparoscopes and Arthroscopes

Although high-level disinfection appears to be the minimum standard for processing laparoscopes and arthroscopes between patients [28, 86, 174, 175], this practice continues to be debated [89, 90, 176]. However, neither side in the high-level disinfection versus sterilization debate has sufficient data on which to base its conclusions. Proponents of high-level disinfection refer to membership surveys [29] or institutional experiences [87] involving more than 117,000 and 10,000 laparoscopic procedures, respectively, that cite a low risk for infection (<0.3%) when high-level disinfection is used for gynecologic laparoscopic equipment. Only one infection in the membership survey was linked to spores. In addition, growth of common skin microorganisms (e.g., *Staphylococcus epidermidis*, diphtheroids) has been documented from the umbilical area even after skin preparation with povidone-iodine and ethyl alcohol. Similar organisms were recovered in some instances from the pelvic serosal surfaces or from the laparoscopic telescopes, suggesting that the microorganisms probably were carried from the skin into the peritoneal cavity [177, 178]. Proponents of sterilization focus on the possibility of transmitting infection by spore-forming organisms. Researchers have proposed several reasons why sterility was not necessary for all laparoscopic equipment: only a limited number of organisms (usually $\leq$10) are introduced into the peritoneal cavity during laparoscopy; minimal damage is done to inner abdominal structures with little devitalized tissue; the peritoneal cavity tolerates small numbers of spore-forming bacteria; equipment is simple to clean and disinfect; surgical sterility is relative; the natural bioburden on rigid lumened devices is low[179]; and no evidence exists that high-level disinfection instead of sterilization increases the risk for infection [87, 89, 90]. With the advent of laparoscopic cholecystectomy, concern about high-level disinfection is justifiable because the degree of tissue damage and bacterial contamination is greater than with laparoscopic procedures in gynecology. Failure to completely dissemble, clean, and high-level disinfect laparoscope parts has led to infections in patients[180]. Data from one study suggested that disassembly, cleaning, and proper reassembly of laparoscopic equipment used in gynecologic procedures before steam sterilization presents no risk for infection[181].

As with laparoscopes and other equipment that enter sterile body sites, arthroscopes ideally should be sterilized before used. Older studies demonstrated that these instruments were commonly (57%) only high-level disinfected in the United States [28, 86]. A later survey (with a response rate of only 5%) reported that high-level disinfection was used by 31% and a sterilization process in the remainder of the health-care facilities[30] High-level disinfection rather than sterilization presumably has been used because the incidence of infection is low and the few infections identified probably are unrelated to the use of high-level disinfection rather than sterilization. A retrospective study of 12,505 arthroscopic procedures found an infection rate of 0.04% (five infections) when arthroscopes were soaked in 2% glutaraldehyde for 15–20 minutes. Four infections were caused by *S. aureus*; the fifth was an anaerobic streptococcal infection [88]. Because these organisms are very susceptible to high-level disinfectants, such as 2% glutaraldehyde, the infections most likely originated from the patient's skin. Two cases of *Clostridium perfringens* arthritis have been reported when the arthroscope was disinfected with glutaraldehyde for an exposure time that is not effective against spores [182, 183].

Although only limited data are available, the evidence does not demonstrate that high-level disinfection of arthroscopes and laparoscopes poses an infection risk to the patient. For example, a prospective study that compared the reprocessing of arthroscopes and laparoscopes (per 1,000 procedures) with EtO sterilization to high-level disinfection with glutaraldehyde found no statistically significant difference in infection risk between the two methods (i.e., EtO, 7.5/1,000 procedures; glutaraldehyde, 2.5/1,000 procedures)[89]. Although the debate for high-level disinfection versus sterilization of laparoscopes and arthroscopes will go unsettled until well-designed, randomized clinical trials are published, this guideline should be followed [1, 17]. That is, laparoscopes, arthroscopes, and other scopes that enter normally sterile tissue should be sterilized before each use; if this is not feasible, they should receive at least high-level disinfection.

Tonometers, Cervical Diaphragm Fitting Rings, Cryosurgical Instruments, and Endocavitary Probes

Disinfection strategies vary widely for other semicritical items (e.g., applanation tonometers, rectal/vaginal probes, cryosurgical instruments, and diaphragm fitting rings). FDA requests that device manufacturers include at least one validated cleaning and disinfection/sterilization protocol in the labeling for their devices. As with all medications and devices, users should be familiar with the label instructions. One study revealed that no uniform technique was in use for disinfection of applanation tonometers, with disinfectant contact times varying from <15 sec to 20 minutes [28]. In view of the potential for transmission of viruses (e.g., herpes simplex virus [HSV], adenovirus 8, or HIV) [184] by tonometer tips, CDC recommended that the tonometer tips be wiped clean and disinfected for 5-10 minutes with either 3% hydrogen peroxide, 5000 ppm chlorine, 70% ethyl alcohol, or 70% isopropyl alcohol [95]. However, more recent data suggest that 3% hydrogen peroxide and 70% isopropyl alcohol are not effective against adenovirus capable of causing epidemic keratoconjunctivitis and similar viruses and should not be used for disinfecting applanation tonometers [49, 185, 186]. Structural damage to Schiotz tonometers has been observed with a 1:10 sodium hypochlorite (5,000 ppm chlorine) and 3% hydrogen peroxide[187]. After disinfection, the tonometer should be thoroughly rinsed in tapwater and air dried before use. Although these disinfectants and exposure times should kill pathogens that can infect the eyes, no studies directly support this [188, 189]. The guidelines of the American Academy of Ophthalmology for preventing infections in ophthalmology focus on only one potential pathogen: HIV. [190] Because a short and simple decontamination procedure is desirable in the clinical setting, swabbing the tonometer tip with a 70% isopropyl alcohol wipe sometimes is practiced. [189] Preliminary reports suggest that wiping the tonometer tip with an alcohol swab and then allowing the alcohol to evaporate might be effective in eliminating HSV, HIV, and adenovirus[189, 191, 192]. However, because these studies involved only a few replicates and were conducted in a controlled laboratory setting, further studies are needed before this technique can be recommended. In addition, two reports have found that disinfection of pneumotonometer tips between uses with a 70% isopropyl alcohol wipe contributed to outbreaks of epidemic keratoconjunctivitis caused

by adenovirus type 8[193, 194].

Limited studies have evaluated disinfection techniques for other items that contact mucous membranes, such as diaphragm fitting rings, cryosurgical probes, transesophageal echocardiography probes [195], flexible cystoscopes [196] or vaginal/rectal probes used in sonographic scanning. Lettau, Bond, and McDougal of CDC supported the recommendation of a diaphragm fitting ring manufacturer that involved using a soap-and-water wash followed by a 15-minute immersion in 70% alcohol[96]. This disinfection method should be adequate to inactivate HIV, HBV, and HSV even though alcohols are not classified as high-level disinfectants because their activity against picornaviruses is somewhat limited[72]. No data are available regarding inactivation of human papillomavirus (HPV) by alcohol or other disinfectants because in vitro replication of complete virions has not been achieved. Thus, even though alcohol for 15 minutes should kill pathogens of relevance in gynecology, no clinical studies directly support this practice.

Vaginal probes are used in sonographic scanning. A vaginal probe and all endocavitary probes without a probe cover are semicritical devices because they have direct contact with mucous membranes (e.g., vagina, rectum, pharynx). While use of the probe cover could be considered as changing the category, this guideline proposes use of a new condom/probe cover for the probe for each patient, and because condoms/probe covers can fail [195, 197-199], the probe also should be high-level disinfected. The relevance of this recommendation is reinforced with the findings that sterile transvaginal ultrasound probe covers have a very high rate of perforations even before use (0%, 25%, and 65% perforations from three suppliers). [199] One study found, after oocyte retrieval use, a very high rate of perforations in used endovaginal probe covers from two suppliers (75% and 81%) [199], other studies demonstrated a lower rate of perforations after use of condoms (2.0% and 0.9%) [197 200]. Condoms have been found superior to commercially available probe covers for covering the ultrasound probe (1.7% for condoms versus 8.3% leakage for probe covers)[201]. These studies underscore the need for routine probe disinfection between examinations. Although most ultrasound manufacturers recommend use of 2% glutaraldehyde for high-level disinfection of contaminated transvaginal transducers, the this agent has been questioned [202] because it might shorten the life of the transducer and might have toxic effects on the gametes and embryos [203]. An alternative procedure for disinfecting the vaginal transducer involves the mechanical removal of the gel from the transducer, cleaning the transducer in soap and water, wiping the transducer with 70% alcohol or soaking it for 2 minutes in 500 ppm chlorine, and rinsing with tap water and air drying[204]. The effectiveness of this and other methods [200] has not been validated in either rigorous laboratory experiments or in clinical use. High-level disinfection with a product (e.g., hydrogen peroxide) that is not toxic to staff, patients, probes, and retrieved cells should be used until the effectiveness of alternative procedures against microbes of importance at the cavitary site is demonstrated by well-designed experimental scientific studies. Other probes such as rectal, cryosurgical, and transesophageal probes or devices also should be high-level disinfected between patients.

Ultrasound probes used during surgical procedures also can contact sterile body sites. These probes can be covered with a sterile sheath to reduce the level of contamination on the probe and reduce the risk for infection. However, because the sheath does not completely protect the probe, the probes should be sterilized between each patient use as with other critical items. If this is not possible, at a minimum the probe should be high-level disinfected and covered with a sterile probe cover.

Some cryosurgical probes are not fully immersible. During reprocessing, the tip of the probe should be immersed in a high-level disinfectant for the appropriate time; any other portion of the probe that could have mucous membrane contact can be disinfected by immersion or by wrapping with a cloth soaked in a high-level disinfectant to allow the recommended contact time. After disinfection, the probe should be rinsed with tap water and dried before use. Health-care facilities that use nonimmersible probes should replace them as soon as possible with fully immersible probes.

As with other high-level disinfection procedures, proper cleaning of probes is necessary to ensure the success of the subsequent disinfection [205]. One study demonstrated that vegetative bacteria

inoculated on vaginal ultrasound probes decreased when the probes were cleaned with a towel [206]. No information is available about either the level of contamination of such probes by potential viral pathogens such as HBV and HPV or their removal by cleaning (such as with a towel). Because these pathogens might be present in vaginal and rectal secretions and contaminate probes during use, high-level disinfection of the probes after such use is recommended.

Dental Instruments

Scientific articles and increased publicity about the potential for transmitting infectious agents in dentistry have focused attention on dental instruments as possible agents for pathogen transmission[207, 208]. The American Dental Association recommends that surgical and other instruments that normally penetrate soft tissue or bone (e.g., extraction forceps, scalpel blades, bone chisels, periodontal scalers, and surgical burs) be classified as critical devices that should be sterilized after each use or discarded. Instruments not intended to penetrate oral soft tissues or bone (e.g., amalgam condensers, and air/water syringes) but that could contact oral tissues are classified as semicritical, but sterilization after each use is recommended if the instruments are heat-tolerant [43, 209]. If a semicritical item is heat–sensitive, it should, at a minimum, be processed with high-level disinfection [43, 210]. Handpieces can be contaminated internally with patient material and should be heat sterilized after each patient. Handpieces that cannot be heat sterilized should not be used. [211] Methods of sterilization that can be used for critical or semicritical dental instruments and materials that are heat-stable include steam under pressure (autoclave), chemical (formaldehyde) vapor, and dry heat (e.g., 320°F for 2 hours). Dental professionals most commonly use the steam sterilizer [212]. All three sterilization procedures can damage some dental instruments, including steam-sterilized hand pieces [213]. Heat-tolerant alternatives are available for most clinical dental applications and are preferred[43].

CDC has divided noncritical surfaces in dental offices into clinical contact and housekeeping surfaces[43]. Clinical contact surfaces are surfaces that might be touched frequently with gloved hands during patient care or that might become contaminated with blood or other potentially infectious material and subsequently contact instruments, hands, gloves, or devices (e.g., light handles, switches, dental X-ray equipment, chair-side computers). Barrier protective coverings (e.g., clear plastic wraps) can be used for these surfaces, particularly those that are difficult to clean (e.g., light handles, chair switches). The coverings should be changed when visibly soiled or damaged and routinely (e.g., between patients). Protected surfaces should be disinfected at the end of each day or if contamination is evident. If not barrier-protected, these surfaces should be disinfected between patients with an intermediate-disinfectant (i.e., EPA-registered hospital disinfectant with tuberculocidal claim) or low-level disinfectant (i.e., EPA-registered hospital disinfectant with an HBV and HIV label claim) [43, 214, 215].

Most housekeeping surfaces need to be cleaned only with a detergent and water or an EPA-registered hospital disinfectant, depending of the nature of the surface and the type and degree of contamination. When housekeeping surfaces are visibly contaminated by blood or body substances, however, prompt removal and surface disinfection is a sound infection control practice and required by the Occupational Safety and Health Administration (OSHA) [43, 214].

Several studies have demonstrated variability among dental practices while trying to meet these recommendations[216, 217]. For example, 68% of respondents believed they were sterilizing their instruments but did not use appropriate chemical sterilants or exposure times and 49% of respondents did not challenge autoclaves with biological indicators[216]. Other investigators using biologic indicators have found a high proportion (15%–65%) of positive spore tests after assessing the efficacy of sterilizers used in dental offices. In one study of Minnesota dental offices, operator error, rather than mechanical malfunction[218], caused 87% of sterilization failures. Common factors in the improper use of sterilizers include chamber overload, low temperature setting, inadequate exposure time, failure to preheat the sterilizer, and interruption of the cycle.

Mail-return sterilization monitoring services use spore strips to test sterilizers in dental clinics, but

delay caused by mailing to the test laboratory could potentially cause false-negatives results. Studies revealed, however, that the post-sterilization time and temperature after a 7-day delay had no influence on the test results[219]. Delays (7 days at 27°C and 37°C, 3-day mail delay) did not cause any predictable pattern of inaccurate spore tests [220].

Disinfection of HBV-, HCV-, HIV- or TB-Contaminated Devices

The CDC recommendation for high-level disinfection of HBV-, HCV-, HIV- or TB-contaminated devices is appropriate because experiments have demonstrated the effectiveness of high-level disinfectants to inactivate these and other pathogens that might contaminate semicritical devices [61, 62, 73, 81, 105, 121, 125, 221-238]. Nonetheless, some healthcare facilities have modified their disinfection procedures when endoscopes are used with a patient known or suspected to be infected with HBV, HIV, or *M. tuberculosis* [28, 239]. This is inconsistent with the concept of Standard Precautions that presumes all patients are potentially infected with bloodborne pathogens[228]. Several studies have highlighted the inability to distinguish HBV- or HIV-infected patients from noninfected patients on clinical grounds[240-242]. In addition, mycobacterial infection is unlikely to be clinically apparent in many patients. In most instances, hospitals that altered their disinfection procedure used EtO sterilization on the endoscopic instruments because they believed this practice reduced the risk for infection [28, 239]. EtO is not routinely used for endoscope sterilization because of the lengthy processing time. Endoscopes and other semicritical devices should be managed the same way regardless of whether the patient is known to be infected with HBV, HCV, HIV or *M. tuberculosis*.

An evaluation of a manual disinfection procedure to eliminate HCV from experimentally contaminated endoscopes provided some evidence that cleaning and 2% glutaraldehyde for 20 minutes should prevent transmission [236]. A study that used experimentally contaminated hysteroscopes detected HCV by polymerase chain reaction (PCR) in one (3%) of 34 samples after cleaning with a detergent, but no samples were positive after treatment with a 2% glutaraldehyde solution for 20 minutes [120]. Another study demonstrated complete elimination of HCV (as detected by PCR) from endoscopes used on chronically infected patients after cleaning and disinfection for 3–5 minutes in glutaraldehyde [118]. Similarly, PCR was used to demonstrate complete elimination of HCV after standard disinfection of experimentally contaminated endoscopes [236] and endoscopes used on HCV-antibody–positive patients had no detectable HCV RNA after high-level disinfection [243]. The inhibitory activity of a phenolic and a chlorine compound on HCV showed that the phenolic inhibited the binding and replication of HCV, but the chlorine was ineffective, probably because of its low concentration and its neutralization in the presence of organic matter [244].

Disinfection in the Hemodialysis Unit

Hemodialysis systems include hemodialysis machines, water supply, water-treatment systems, and distribution systems. During hemodialysis, patients have acquired bloodborne viruses and pathogenic bacteria [245-247]. Cleaning and disinfection are important components of infection control in a hemodialysis center. EPA and FDA regulate disinfectants used to reprocess hemodialyzers, hemodialysis machines, and water-treatment systems.

Noncritical surfaces (e.g., dialysis bed or chair, countertops, external surfaces of dialysis machines, and equipment [scissors, hemostats, clamps, blood pressure cuffs, stethoscopes]) should be disinfected with an EPA-registered disinfectant unless the item is visibly contaminated with blood; in that case a tuberculocidal agent (or a disinfectant with specific label claims for HBV and HIV) or a 1:100 dilution of a hypochlorite solution (500–600 ppm free chlorine) should be used [246, 248]. This procedure accomplishes two goals: it removes soil on a regular basis and maintains an environment that is consistent with good patient care. Hemodialyzers are disinfected with peracetic acid, formaldehyde, glutaraldehyde, heat pasteurization with citric acid, and chlorine-containing compounds [249]. Hemodialysis systems usually are disinfected by chlorine-based disinfectants (e.g., sodium hypochlorite), aqueous

formaldehyde, heat pasteurization, ozone, or peracetic acid [250, 251]. All products must be used according to the manufacturers' recommendations. Some dialysis systems use hot-water disinfection to control microbial contamination.

At its high point, 82% of U.S. chronic hemodialysis centers were reprocessing (i.e., reusing) dialyzers for the same patient using high-level disinfection [249]. However, one of the large dialysis organizations has decided to phase out reuse and, by 2002 the percentage of dialysis facilities reprocessing hemodialyzers had decreased to 63% [252]. The two commonly used disinfectants to reprocess dialyzers were peracetic acid and formaldehyde; 72% used peracetic acid and 20% used formaldehyde to disinfect hemodialyzers. Another 4% of the facilities used either glutaraldehyde or heat pasteurization in combination with citric acid [252]. Infection-control recommendations, including disinfection and sterilization and the use of dedicated machines for hepatitis B surface antigen (HBsAg)-positive patients, in the hemodialysis setting were detailed in two reviews [245, 246]. The Association for the Advancement of Medical Instrumentation(AAMI) has published recommendations for the reuse of hemodialyzers[253].

Inactivation of *Clostridium difficile*

The source of health-care–associated acquisition of *Clostridium difficile* in nonepidemic settings has not been determined. The environment and carriage on the hands of health-care personnel have been considered possible sources of infection [66, 254]. Carpeted rooms occupied by a patient with *C. difficile* were more heavily contaminated with *C. difficile* than were noncarpeted rooms [255]. Because *C. difficile* spore-production can increase when exposed to nonchlorine-based cleaning agents and the spores are more resistant than vegetative cells to commonly used surface disinfectants[256], some investigators have recommended use of dilute solutions of hypochlorite (1,600 ppm available chlorine) for routine environmental disinfection of rooms of patients with *C. difficile*-associated diarrhea or colitis [257], to reduce the incidence of *C. difficile* diarrhea [258], or in units with high *C. difficile* rates. [259] Stool samples of patients with symptomatic *C. difficile* colitis contain spores of the organism, as demonstrated by ethanol treatment of the stool to reduce the overgrowth of fecal flora when isolating *C. difficile* in the laboratory[260, 261]. *C. difficile*-associated diarrhea rates were shown to have decreased markedly in a bone-marrow transplant unit (from 8.6 to 3.3 cases per 1,000 patient-days) during a period of bleach disinfection (1:10 dilution) of environmental surfaces compared with cleaning with a quaternary ammonium compound. Because no EPA-registered products exist that are specific for inactivating *C. difficile* spores, use of diluted hypochlorite should be considered in units with high *C. difficile* rates. Acidified bleach and regular bleach (5000 ppm chlorine) can inactivate 10^6 *C. difficile* spores in $\leq$10 minutes [262]. However, studies have shown that asymptomatic patients constitute an important reservoir within the health-care facility and that person-to-person transmission is the principal means of transmission between patients. Thus, combined use of hand washing, barrier precautions, and meticulous environmental cleaning with an EPA-registered disinfectant (e.g., germicidal detergent) should effectively prevent spread of the organism [263].

Contaminated medical devices, such as colonoscopes and thermometers,can be vehicles for transmission of *C. difficile* spores [264]. For this reason, investigators have studied commonly used disinfectants and exposure times to assess whether current practices can place patients at risk. Data demonstrate that 2% glutaraldehyde [79, 265-267] and peracetic acid [267, 268] reliably kill *C. difficile* spores using exposure times of 5–20 minutes. *ortho*-Phthalaldehyde and $\geq$0.2% peracetic acid (WA Rutala, personal communication, April 2006) also can inactivate $\geq 10^4$ *C. difficile* spores in 10–12 minutes at 20°C [268]. Sodium dichloroisocyanurate at a concentration of 1000 ppm available chlorine achieved lower $\log_{10}$ reduction factors against *C. difficile* spores at 10 min, ranging from 0.7 to 1.5, than 0.26% peracetic acid with $\log_{10}$ reduction factors ranging from 2.7 to 6.0[268].

OSHA Bloodborne Pathogen Standard

In December 1991, OSHA promulgated a standard entitled "Occupational Exposure to

Bloodborne Pathogens" to eliminate or minimize occupational exposure to bloodborne pathogens [214]. One component of this requirement is that all equipment and environmental and working surfaces be cleaned and decontaminated with an appropriate disinfectant after contact with blood or other potentially infectious materials. Even though the OSHA standard does not specify the type of disinfectant or procedure, the OSHA original compliance document [269] suggested that a germicide must be tuberculocidal to kill the HBV. To follow the OSHA compliance document a tuberculocidal disinfectant (e.g., phenolic, and chlorine) would be needed to clean a blood spill. However, in February 1997, OSHA amended its policy and stated that EPA-registered disinfectants labeled as effective against HIV and HBV would be considered as appropriate disinfectants ". . . provided such surfaces have not become contaminated with agent(s) or volumes of or concentrations of agent(s) for which higher level disinfection is recommended." When bloodborne pathogens other than HBV or HIV are of concern, OSHA continues to require use of EPA-registered tuberculocidal disinfectants or hypochlorite solution (diluted 1:10 or 1:100 with water) [215, 228]. Studies demonstrate that, in the presence of large blood spills, a 1:10 final dilution of EPA-registered hypochlorite solution initially should be used to inactivate bloodborne viruses [63, 235] to minimize risk for infection to health-care personnel from percutaneous injury during cleanup.

Emerging Pathogens (*Cryptosporidium, Helicobacter pylori, Escherichia coli* O157:H7, Rotavirus, Human Papilloma Virus, Norovirus, Severe Acute Respiratory Syndrome [SARS] Coronavirus)

Emerging pathogens are of growing concern to the general public and infection-control professionals. Relevant pathogens include *Cryptosporidium parvum, Helicobacter pylori, E. coli* O157:H7, HIV, HCV, rotavirus, norovirus, severe acute respiratory syndrome (SARS) coronavirus, multidrug-resistant *M. tuberculosis*, and nontuberculous mycobacteria (e.g., *M. chelonae*). The susceptibility of each of these pathogens to chemical disinfectants and sterilants has been studied. With the exceptions discussed below, all of these emerging pathogens are susceptible to currently available chemical disinfectants and sterilants [270].

Cryptosporidium is resistant to chlorine at concentrations used in potable water. *C. parvum* is not completely inactivated by most disinfectants used in healthcare including ethyl alcohol [271], glutaraldehyde [271, 272], 5.25% hypochlorite [271], peracetic acid [271], ortho-phthalaldehyde [271], phenol [271, 272], povidone-iodine [271, 272], and quaternary ammonium compounds[271]. The only chemical disinfectants and sterilants able to inactivate greater than 3 $\log_{10}$ of *C. parvum* were 6% and 7.5% hydrogen peroxide [271]. Sterilization methods will fully inactivate *C. parvum,* including steam [271], EtO [271, 273], and hydrogen peroxide gas plasma[271]. Although most disinfectants are ineffective against *C. parvum*, current cleaning and disinfection practices appear satisfactory to prevent healthcare-associated transmission. For example, endoscopes are unlikely to be an important vehicle for transmitting *C. parvum* because the results of bacterial studies indicate mechanical cleaning will remove approximately 10^4 organisms, and drying results in rapid loss of *C. parvum* viability (e.g., 30 minutes, 2.9 $\log_{10}$ decrease; and 60 minutes, 3.8 $\log_{10}$ decrease) [271].

Chlorine at ~1 ppm has been found capable of eliminating approximately 4 $\log_{10}$ of *E. coli* O157:H7 within 1 minute in a suspension test[64]. Electrolyzed oxidizing water at 23°C was effective in 10 minutes in producing a 5-$\log_{10}$ decrease in *E. coli* O157:H7 inoculated onto kitchen cutting boards[274]. The following disinfectants eliminated >5 $\log_{10}$ of *E. coli* O157:H7 within 30 seconds: a quaternary ammonium compound, a phenolic, a hypochlorite (1:10 dilution of 5.25% bleach), and ethanol[53]. Disinfectants including chlorine compounds can reduce *E. coli* O157:H7 experimentally inoculated onto alfalfa seeds or sprouts [275, 276] or beef carcass surfaces[277].

Data are limited on the susceptibility of *H. pylori* to disinfectants. Using a suspension test, one study assessed the effectiveness of a variety of disinfectants against nine strains of *H. pylori* [60]. Ethanol (80%) and glutaraldehyde (0.5%) killed all strains within 15 seconds; chlorhexidine gluconate (0.05%, 1.0%), benzalkonium chloride (0.025%, 0.1%), alkyldiaminoethylglycine hydrochloride (0.1%), povidone-iodine (0.1%), and sodium hypochlorite (150 ppm) killed all strains within 30 seconds. Both ethanol

(80%) and glutaraldehyde (0.5%) retained similar bactericidal activity in the presence of organic matter; the other disinfectants showed reduced bactericidal activity. In particular, the bactericidal activity of povidone-iodine (0.1%) and sodium hypochlorite (150 ppm) markedly decreased in the presence of dried yeast solution with killing times increased to 5 - 10 minutes and 5 - 30 minutes, respectively.

Immersing biopsy forceps in formalin before obtaining a specimen does not affect the ability to culture *H. pylori* from the biopsy specimen [278]. The following methods are ineffective for eliminating *H. pylori* from endoscopes: cleaning with soap and water [119, 279], immersion in 70% ethanol for 3 minutes[280], instillation of 70% ethanol[126], instillation of 30 ml of 83% methanol[279], and instillation of 0.2% Hyamine solution[281]. The differing results with regard to the efficacy of ethyl alcohol against *Helicobacter* are unexplained. Cleaning followed by use of 2% alkaline glutaraldehyde (or automated peracetic acid) has been demonstrated by culture to be effective in eliminating *H. pylori* [119, 279, 282]. Epidemiologic investigations of patients who had undergone endoscopy with endoscopes mechanically washed and disinfected with 2.0%–2.3% glutaraldehyde have revealed no evidence of person-to-person transmission of *H. pylori* [126, 283]. Disinfection of experimentally contaminated endoscopes using 2% glutaraldehyde (10-minute, 20-minute, 45-minute exposure times) or the peracetic acid system (with and without active peracetic acid) has been demonstrated to be effective in eliminating *H. pylori* [119]. *H. pylori* DNA has been detected by PCR in fluid flushed from endoscope channels after cleaning and disinfection with 2% glutaraldehyde [284]. The clinical significance of this finding is unclear. *In vitro* experiments have demonstrated a >3.5-$\log_{10}$ reduction in *H. pylori* after exposure to 0.5 mg/L of free chlorine for 80 seconds[285].

An outbreak of healthcare-associated rotavirus gastroenteritis on a pediatric unit has been reported [286]. Person to person through the hands of health-care workers was proposed as the mechanism of transmission. Prolonged survival of rotavirus on environmental surfaces (90 minutes to >10 days at room temperature) and hands (>4 hours) has been demonstrated. Rotavirus suspended in feces can survive longer [287, 288]. Vectors have included hands, fomites, air, water, and food [288, 289]. Products with demonstrated efficacy (>3 $\log_{10}$ reduction in virus) against rotavirus within 1 minute include: 95% ethanol, 70% isopropanol, some phenolics, 2% glutaraldehyde, 0.35% peracetic acid, and some quaternary ammonium compounds [59, 290-293]. In a human challenge study, a disinfectant spray (0.1% ortho-phenylphenol and 79% ethanol), sodium hypochlorite (800 ppm free chlorine), and a phenol-based product (14.7% phenol diluted 1:256 in tapwater) when sprayed onto contaminated stainless steel disks**,** were effective in interrupting transfer of a human rotavirus from stainless steel disk to fingerpads of volunteers after an exposure time of 3- 10 minutes. A quaternary ammonium product (7.05% quaternary ammonium compound diluted 1:128 in tapwater) and tapwater allowed transfer of virus [52].

No data exist on the inactivation of HPV by alcohol or other disinfectants because in vitro replication of complete virions has not been achieved. Similarly, little is known about inactivation of noroviruses (members of the family *Caliciviridae* and important causes of gastroenteritis in humans) because they cannot be grown in tissue culture. Improper disinfection of environmental surfaces contaminated by feces or vomitus of infected patients is believed to play a role in the spread of noroviruses in some settings [294-296]. Prolonged survival of a norovirus surrogate (i.e., feline calicivirus virus [FCV], a closely related cultivable virus) has been demonstrated (e.g., at room temperature, FCV in a dried state survived for 21–18 days) [297]. Inactivation studies with FCV have shown the effectiveness of chlorine, glutaraldehyde, and iodine-based products whereas the quaternary ammonium compound, detergent, and ethanol failed to inactivate the virus completely. [297] An evaluation of the effectiveness of several disinfectants against the feline calicivirus found that bleach diluted to 1000 ppm of available chlorine reduced infectivity of FCV by 4.5 logs in 1 minute. Other effective ($\log_{10}$ reduction factor of >4 in virus) disinfectants included accelerated hydrogen peroxide, 5,000 ppm (3 min); chlorine dioxide, 1,000 ppm chlorine (1 min); a mixture of four quaternary ammonium compounds, 2,470 ppm (10 min); 79% ethanol with 0.1% quaternary ammonium compound (3 min); and 75% ethanol (10 min) [298]. A quaternary ammonium compound exhibited activity against feline calicivirus supensions dried on hard surface carriers in 10 minutes [299]. Seventy percent ethanol and 70% 1-propanol reduced FCV by a 3–4-$\log_{10}$

reduction in 30 seconds [300].

CDC announced that a previously unrecognized human virus from the coronavirus family is the leading hypothesis for the cause of a described syndrome of SARS [301]. Two coronaviruses that are known to infect humans cause one third of common colds and can cause gastroenteritis. The virucidal efficacy of chemical germicides against coronavirus has been investigated. A study of disinfectants against coronavirus 229E found several that were effective after a 1-minute contact time; these included sodium hypochlorite (at a free chlorine concentration of 1,000 ppm and 5,000 ppm), 70% ethyl alcohol, and povidone-iodine (1% iodine) [186]. In another study, 70% ethanol, 50% isopropanol, 0.05% benzalkonium chloride, 50 ppm iodine in iodophor, 0.23% sodium chlorite, 1% cresol soap and 0.7% formaldehyde inactivated >3 logs of two animal coronaviruses (mouse hepatitis virus, canine coronavirus) after a 10-minute exposure time [302]. The activity of povidone-iodine has been demonstrated against human coronaviruses 229E and OC43 [303]. A study also showed complete inactivation of the SARS coronavirus by 70% ethanol and povidone-iodine with an exposure times of 1 minute and 2.5% glutaraldehyde with an exposure time of 5 minute [304]. Because the SARS coronavirus is stable in feces and urine at room temperature for at least 1–2 days (WHO, 2003; http://www.who.int/csr/sars/survival_2003_05_04/en/index.html), surfaces might be a possible source of contamination and lead to infection with the SARS coronavirus and should be disinfected. Until more precise information is available, environments in which SARS patients are housed should be considered heavily contaminated, and rooms and equipment should be thoroughly disinfected daily and after the patient is discharged. EPA-registered disinfectants or 1:100 dilution of household bleach and water should be used for surface disinfection and disinfection on noncritical patient-care equipment. High-level disinfection and sterilization of semicritical and critical medical devices, respectively, does not need to be altered for patients with known or suspected SARS.

Free-living amoeba can be pathogenic and can harbor agents of pneumonia such as *Legionella pneumophila*. Limited studies have shown that 2% glutaraldehyde and peracetic acid do not completely inactivate *Acanthamoeba polyphaga* in a 20-minute exposure time for high-level disinfection. If amoeba are found to contaminate instruments and facilitate infection, longer immersion times or other disinfectants may need to be considered [305].

Inactivation of Bioterrorist Agents

Publications have highlighted concerns about the potential for biological terrorism[306, 307]. CDC has categorized several agents as “high priority” because they can be easily disseminated or transmitted from person to person, cause high mortality, and are likely to cause public panic and social disruption [308]. These agents include *Bacillus anthracis* (the cause of anthrax), *Yersinia pestis* (plague), variola major (smallpox), *Clostridium botulinum* toxin (botulism), *Francisella tularensis* (tularemia), filoviruses (Ebola hemorrhagic fever, Marburg hemorrhagic fever); and arenaviruses (Lassa [Lassa fever], Junin [Argentine hemorrhagic fever]), and related viruses[308].

A few comments can be made regarding the role of sterilization and disinfection of potential agents of bioterrorism[309]. First, the susceptibility of these agents to germicides *in vitro* is similar to that of other related pathogens. For example, variola is similar to vaccinia [72, 310, 311] and *B. anthracis* is similar to *B. atrophaeus* (formerly *B. subtilis)*[312, 313]. *B. subtilis* spores, for instance, proved as resistant as, if not more resistant than, *B. anthracis* spores (>6 $\log_{10}$ reduction of *B. anthracis* spores in 5 minutes with acidified bleach [5,250 ppm chlorine])[313]. Thus, one can extrapolate from the larger database available on the susceptibility of genetically similar organisms[314]. Second, many of the potential bioterrorist agents are stable enough in the environment that contaminated environmental surfaces or fomites could lead to transmission of agents such as *B. anthracis*, *F. tularensis*, variola major, *C. botulinum* toxin, and *C. burnetti* [315]. Third, data suggest that current disinfection and sterilization practices are appropriate for managing patient-care equipment and environmental surfaces when potentially contaminated patients are evaluated and/or admitted in a health-care facility after exposure to a bioterrorist agent. For example,

sodium hypochlorite can be used for surface disinfection (see http://www.epa.gov/pesticides/factsheets/chemicals/bleachfactsheet.htm). In instances where the health-care facility is the site of a bioterrorist attack, environmental decontamination might require special decontamination procedures (e.g., chlorine dioxide gas for *B. anthracis* spores). Because no antimicrobial products are registered for decontamination of biologic agents after a bioterrorist attack, EPA has granted a crises exemption for each product (see http://www.epa.gov/pesticides/factsheets/chemicals/bleachfactsheet.htm). Of only theoretical concern is the possibility that a bioterrorist agent could be engineered to be less susceptible to disinfection and sterilization processes [309].

Toxicological, Environmental and Occupational Concerns

Health hazards associated with the use of germicides in healthcare vary from mucous membrane irritation to death, with the latter involving accidental injection by mentally disturbed patients[316]. Although their degrees of toxicity vary [317-320], all disinfectants should be used with the proper safety precautions [321] and only for the intended purpose.

Key factors associated with assessing the health risk of a chemical exposure include the duration, intensity (i.e., how much chemical is involved), and route (e.g., skin, mucous membranes, and inhalation) of exposure. Toxicity can be acute or chronic. Acute toxicity usually results from an accidental spill of a chemical substance. Exposure is sudden and often produces an emergency situation. Chronic toxicity results from repeated exposure to low levels of the chemical over a prolonged period. Employers are responsible for informing workers about the chemical hazards in the workplace and implementing control measures. The OSHA Hazard Communication Standard (29 CFR 1910.1200, 1915.99, 1917.28, 1918.90, 1926.59, and 1928.21) requires manufacturers and importers of hazardous chemicals to develop Material Safety Data Sheets (MSDS) for each chemical or mixture of chemicals. Employers must have these data sheets readily available to employees who work with the products to which they could be exposed.

Exposure limits have been published for many chemicals used in health care to help provide a safe environment and, as relevant, are discussed in each section of this guideline. Only the exposure limits published by OSHA carry the legal force of regulations. OSHA publishes a limit as a time-weighted average (TWA), that is, the average concentration for a normal 8-hour work day and a 40-hour work week to which nearly all workers can be repeatedly exposed to a chemical without adverse health effects. For example, the permissible exposure limit (PEL) for EtO is 1.0 ppm, 8 hour TWA. The CDC National Institute for Occupational Safety and Health (NIOSH) develops recommended exposure limits (RELs). RELs are occupational exposure limits recommended by NIOSH as being protective of worker health and safety over a working lifetime. This limit is frequently expressed as a 40-hour TWA exposure for up to 10 hours per day during a 40-hour work week. These exposure limits are designed for inhalation exposures. Irritant and allergic effects can occur below the exposure limits, and skin contact can result in dermal effects or systemic absorption without inhalation. The American Conference on Governmental Industrial Hygienists (ACGIN) also provides guidelines on exposure limits [322]. Information about workplace exposures and methods to reduce them (e.g., work practices, engineering controls, PPE) is available on the OSHA (http://www.osha.gov) and NIOSH (http://www.cdc.gov/niosh) websites.

Some states have excluded or limited concentrations of certain chemical germicides (e.g., glutaraldehyde, formaldehyde, and some phenols) from disposal through the sewer system. These rules are intended to minimize environmental harm. If health-care facilities exceed the maximum allowable concentration of a chemical (e.g., $\geq$5.0 mg/L), they have three options. First, they can switch to alternative products; for example, they can change from glutaraldehyde to another disinfectant for high-level disinfection or from phenolics to quaternary ammonium compounds for low-level disinfection. Second, the health-care facility can collect the disinfectant and dispose of it as a hazardous chemical. Third, the

facility can use a commercially available small-scale treatment method (e.g., neutralize glutaraldehyde with glycine).

Safe disposal of regulated chemicals is important throughout the medical community. For disposal of large volumes of spent solutions, users might decide to neutralize the microbicidal activity before disposal (e.g., glutaraldehyde). Solutions can be neutralized by reaction with chemicals such as sodium bisulfite [323, 324] or glycine [325].

European authors have suggested that instruments and ventilation therapy equipment should be disinfected by heat rather than by chemicals. The concerns for chemical disinfection include toxic side effects for the patient caused by chemical residues on the instrument or object, occupational exposure to toxic chemicals, and recontamination by rinsing the disinfectant with microbially contaminated tap water [326].

Disinfection in Ambulatory Care, Home Care, and the Home

With the advent of managed healthcare, increasing numbers of patients are now being cared for in ambulatory-care and home settings. Many patients in these settings might have communicable diseases, immunocompromising conditions, or invasive devices. Therefore, adequate disinfection in these settings is necessary to provide a safe patient environment. Because the ambulatory-care setting (i.e., outpatient facility) provides the same risk for infection as the hospital, the Spaulding classification scheme described in this guideline should be followed (Table 1) [17].

The home environment should be much safer than hospitals or ambulatory care. Epidemics should not be a problem, and cross-infection should be rare. The healthcare provider is responsible for providing the responsible family member information about infection-control procedures to follow in the home, including hand hygiene, proper cleaning and disinfection of equipment, and safe storage of cleaned and disinfected devices. Among the products recommended for home disinfection of reusable objects are bleach, alcohol, and hydrogen peroxide. APIC recommends that reusable objects (e.g., tracheostomy tubes) that touch mucous membranes be disinfected by immersion in 70% isopropyl alcohol for 5 minutes or in 3% hydrogen peroxide for 30 minutes. Additionally, a 1:50 dilution of 5.25%–6.15% sodium hypochlorite (household bleach) for 5 minutes should be effective [327-329]. Noncritical items (e.g., blood pressure cuffs, crutches) can be cleaned with a detergent. Blood spills should be handled according to OSHA regulations as previously described (see section on OSHA Bloodborne Pathogen Standard). In general, sterilization of critical items is not practical in homes but theoretically could be accomplished by chemical sterilants or boiling. Single-use disposable items can be used or reusable items sterilized in a hospital [330, 331].

Some environmental groups advocate “environmentally safe” products as alternatives to commercial germicides in the home-care setting. These alternatives (e.g., ammonia, baking soda, vinegar, Borax, liquid detergent) are not registered with EPA and should not be used for disinfecting because they are ineffective against *S. aureus*. Borax, baking soda, and detergents also are ineffective against *Salmonella* Typhi and *E.coli*; however, undiluted vinegar and ammonia are effective against *S.* Typhi and *E.coli* [53, 332, 333]. Common commercial disinfectants designed for home use also are effective against selected antibiotic-resistant bacteria [53].

Public concerns have been raised that the use of antimicrobials in the home can promote development of antibiotic-resistant bacteria [334, 335]. This issue is unresolved and needs to be considered further through scientific and clinical investigations. The public health benefits of using disinfectants in the home are unknown. However, some facts are known: many sites in the home kitchen and bathroom are microbially contaminated [336], use of hypochlorites markedly reduces bacteria [337], and good standards of hygiene (e.g., food hygiene, hand hygiene) can help reduce infections in the home [338, 339]. In addition, laboratory studies indicate that many commercially prepared household disinfectants are effective against common pathogens [53] and can interrupt surface-to-human transmission of pathogens [48]. The “targeted

hygiene concept"—which means identifying situations and areas (e.g., food-preparation surfaces and bathroom) where risk exists for transmission of pathogens—may be a reasonable way to identify when disinfection might be appropriate [340].

Susceptibility of Antibiotic-Resistant Bacteria to Disinfectants

As with antibiotics, reduced susceptibility (or acquired "resistance") of bacteria to disinfectants can arise by either chromosomal gene mutation or acquisition of genetic material in the form of plasmids or transposons [338, 341-343, 344 , 345, 346]. When changes occur in bacterial susceptibility that renders an antibiotic ineffective against an infection previously treatable by that antibiotic, the bacteria are referred to as "resistant." In contrast, reduced susceptibility to disinfectants does not correlate with failure of the disinfectant because concentrations used in disinfection still greatly exceed the cidal level. Thus, the word "resistance" when applied to these changes is incorrect, and the preferred term is "reduced susceptibility" or "increased tolerance"[344, 347]. No data are available that show that antibiotic-resistant bacteria are less sensitive to the liquid chemical germicides than antibiotic-sensitive bacteria at currently used germicide contact conditions and concentrations.

MRSA and vancomycin-resistant *Enterococcus* (VRE) are important health-care–associated agents. Some antiseptics and disinfectants have been known for years to be, because of MICs, somewhat less inhibitory to *S. aureus* strains that contain a plasmid-carrying gene encoding resistance to the antibiotic gentamicin [344]. For example, gentamicin resistance has been shown to also encode reduced susceptibility to propamidine, quaternary ammonium compounds, and ethidium bromide [348], and MRSA strains have been found to be less susceptible than methicillin-sensitive *S. aureus* (MSSA) strains to chlorhexidine, propamidine, and the quaternary ammonium compound cetrimide [349]. In other studies, MRSA and MSSA strains have been equally sensitive to phenols and chlorhexidine, but MRSA strains were slightly more tolerant to quaternary ammonium compounds [350]. Two gene families (*qac*CD [now referred to as *smr*] and *qac*AB) are involved in providing protection against agents that are components of disinfectant formulations such as quaternary ammonium compounds. Staphylococci have been proposed to evade destruction because the protein specified by the *qac*A determinant is a cytoplasmic-membrane–associated protein involved in an efflux system that actively reduces intracellular accumulation of toxicants, such as quaternary ammonium compounds, to intracellular targets [351].

Other studies demonstrated that plasmid-mediated formaldehyde tolerance is transferable from *Serratia marcescens* to *E. coli* [352] and plasmid-mediated quaternary ammonium tolerance is transferable from *S. aureus* to *E. coli*.[353]. Tolerance to mercury and silver also is plasmid borne [341, 343-346].

Because the concentrations of disinfectants used in practice are much higher than the MICs observed, even for the more tolerant strains, the clinical relevance of these observations is questionable. Several studies have found antibiotic-resistant hospital strains of common healthcare-associated pathogens (i.e., *Enterococcus, P. aeruginosa, Klebsiella pneumoniae, E. coli*, *S. aureus,* and *S. epidermidis*) to be equally susceptible to disinfectants as antibiotic-sensitive strains [53, 354-356]. The susceptibility of glycopeptide-intermediate *S. aureus* was similar to vancomycin-susceptible, MRSA [357]. On the basis of these data, routine disinfection and housekeeping protocols do not need to be altered because of antibiotic resistance provided the disinfection method is effective [358, 359]. A study that evaluated the efficacy of selected cleaning methods (e.g., QUAT-sprayed cloth, and QUAT-immersed cloth) for eliminating VRE found that currently used disinfection processes most likely are highly effective in eliminating VRE. However, surface disinfection must involve contact with all contaminated surfaces [358]. A new method using an invisible flurorescent marker to objectively evaluate the thoroughness of cleaning activities in patient rooms might lead to improvement in cleaning of all objects and surfaces but needs further evaluation [360].

Lastly, does the use of antiseptics or disinfectants facilitate the development of disinfectant-tolerant organisms? Evidence and reviews indicate enhanced tolerance to disinfectants can be

developed in response to disinfectant exposure [334, 335, 346, 347, 361]. However, the level of tolerance is not important in clinical terms because it is low and unlikely to compromise the effectiveness of disinfectants of which much higher concentrations are used [347, 362].

The issue of whether low-level tolerance to germicides selects for antibiotic-resistant strains is unsettled but might depend on the mechanism by which tolerance is attained. For example, changes in the permeability barrier or efflux mechanisms might affect susceptibility to both antibiotics and germicides, but specific changes to a target site might not. Some researchers have suggested that use of disinfectants or antiseptics (e.g., triclosan) could facilitate development of antibiotic-resistant microorganisms [334, 335, 363]. Although evidence in laboratory studies indicates low-level resistance to triclosan, the concentrations of triclosan in these studies were low (generally <1 µg/mL) and dissimilar from the higher levels used in antimicrobial products (2,000–20,000 µg/mL) [364, 365]. Thus, researchers can create laboratory-derived mutants that demonstrate reduced susceptibility to antiseptics or disinfectants. In some experiments, such bacteria have demonstrated reduced susceptibility to certain antibiotics [335]. There is no evidence that using antiseptics or disinfectants selects for antibiotic-resistant organisms in nature or that such mutants survive in nature[366].). In addition, the action of antibiotics and the action of disinfectants differ fundamentally. Antibiotics are selectively toxic and generally have a single target site in bacteria, thereby inhibiting a specific biosynthetic process. Germicides generally are considered nonspecific antimicrobials because of a multiplicity of toxic-effect mechanisms or target sites and are broader spectrum in the types of microorganisms against which they are effective [344, 347].

The rotational use of disinfectants in some environments (e.g., pharmacy production units) has been recommended and practiced in an attempt to prevent development of resistant microbes [367, 368]. There have been only rare case reports that appropriately used disinfectants have resulted in a clinical problem arising from the selection or development of nonsusceptible microorganisms [369].

Surface Disinfection

Is Surface Disinfection Necessary?

The effective use of disinfectants is part of a multibarrier strategy to prevent health-care–associated infections. Surfaces are considered noncritical items because they contact intact skin. Use of noncritical items or contact with noncritical surfaces carries little risk of causing an infection in patients or staff. Thus, the routine use of germicidal chemicals to disinfect hospital floors and other noncritical items is controversial [370-375]. A 1991 study expanded the Spaulding scheme by dividing the noncritical environmental surfaces into housekeeping surfaces and medical equipment surfaces [376]. The classes of disinfectants used on housekeeping and medical equipment surfaces can be similar. However, the frequency of decontaminating can vary (see Recommendations). Medical equipment surfaces (e.g., blood pressure cuffs, stethoscopes, hemodialysis machines, and X-ray machines) can become contaminated with infectious agents and contribute to the spread of health-care–associated infections [248, 375]. For this reason, noncritical medical equipment surfaces should be disinfected with an EPA-registered low- or intermediate-level disinfectant. Use of a disinfectant will provide antimicrobial activity that is likely to be achieved with minimal additional cost or work.

Environmental surfaces (e.g., bedside table) also could potentially contribute to cross-transmission by contamination of health-care personnel from hand contact with contaminated surfaces, medical equipment, or patients [50, 375, 377]. A paper reviews the epidemiologic and microbiologic data (Table 3) regarding the use of disinfectants on noncritical surfaces [378].

Of the seven reasons to usie a disinfectant on noncritical surfaces, five are particularly noteworthy and support the use of a germicidal detergent. First, hospital floors become contaminated with microorganisms from settling airborne bacteria: by contact with shoes, wheels, and other objects; and occasionally by spills. The removal of microbes is a component in controling health-care–associated infections. In an investigation of the cleaning of hospital floors, the use of soap and water (80% reduction) was less effective in reducing the numbers of bacteria than was a phenolic disinfectant (94%–99.9%

reduction) [379]. However, a few hours after floor disinfection, the bacterial count was nearly back to the pretreatment level. Second, detergents become contaminated and result in seeding the patient's environment with bacteria. Investigators have shown that mop water becomes increasingly dirty during cleaning and becomes contaminated if soap and water is used rather than a disinfectant. For example, in one study, bacterial contamination in soap and water without a disinfectant increased from 10 CFU/mL to 34,000 CFU/mL after cleaning a ward, whereas contamination in a disinfectant solution did not change (20 CFU/mL) [380]. Contamination of surfaces close to the patient that are frequently touched by the patient or staff (e.g., bed rails) could result in patient exposures0 [381]. In a study, using of detergents on floors and patient room furniture, increased bacterial contamination of the patients' environmental surfaces was found after cleaning (average increase = 103.6 CFU/24cm^2) [382]. In addition, a *P. aeruginosa* outbreak was reported in a hematology-oncology unit associated with contamination of the surface cleaning equipment when nongermicidal cleaning solutions instead of disinfectants were used to decontaminate the patients' environment [383] and another study demonstrated the role of environmental cleaning in controlling an outbreak of *Acinetobacter baumannii* [384]. Studies also have shown that, in situations where the cleaning procedure failed to eliminate contamination from the surface and the cloth is used to wipe another surface, the contamination is transferred to that surface and the hands of the person holding the cloth[381, 385]. Third, the CDC Isolation Guideline recommends that noncritical equipment contaminated with blood, body fluids, secretions, or excretions be cleaned and disinfected after use. The same guideline recommends that, in addition to cleaning, disinfection of the bedside equipment and environmental surfaces (e.g., bedrails, bedside tables, carts, commodes, door-knobs, and faucet handles) is indicated for certain pathogens, e.g., enterococci, which can survive in the inanimate environment for prolonged periods [386]. Fourth, OSHA requires that surfaces contaminated with blood and other potentially infectious materials (e.g., amniotic, pleural fluid) be disinfected. Fifth, using a single product throughout the facility can simplify both training and appropriate practice.

Reasons also exist for using a detergent alone on floors because noncritical surfaces contribute minimally to endemic health-care–associated infections [387], and no differences have been found in healthcare–associated infections rates when floors are cleaned with detergent rather than disinfectant [382, 388, 389]. However, these studies have been small and of short duration and suffer from low statistical power because the outcome—healthcare–associated infections—is of low frequency. The low rate of infections makes the efficacy of an intervention statistically difficult to demonstrate. Because housekeeping surfaces are associated with the lowest risk for disease transmission, some researchers have suggested that either detergents or a disinfectant/detergent could be used [376]. No data exist that show reduced healthcare–associated infection rates with use of surface disinfection of floors, but some data demonstrate reduced microbial load associated with the use of disinfectants. Given this information; other information showing that environmental surfaces (e.g., bedside table, bed rails) close to the patient and in outpatient settings [390] can be contaminated with epidemiologically important microbes (such as VRE and MRSA)[47, 390-394]; and data showing these organisms survive on various hospital surfaces [395, 396]; some researchers have suggested that such surfaces should be disinfected on a regular schedule [378]. Spot decontamination on fabrics that remain in hospitals or clinic rooms while patients move in and out (e.g., privacy curtains) also should be considered. One study demonstrated the effectiveness of spraying the fabric with 3% hydrogen peroxide [397]. Future studies should evaluate the level of contamination on noncritical environmental surfaces as a function of high and low hand contact and whether some surfaces (e.g., bed rails) near the patient with high contact frequencies require more frequent disinfection. Regardless of whether a detergent or disinfectant is used on surfaces in a health-care facility, surfaces should be cleaned routinely and when dirty or soiled to provide an aesthetically pleasing environment and to prevent potentially contaminated objects from serving as a source for health-care–associated infections [398]. The value of designing surfaces (e.g. hexyl-polyvinylpyridine) that kill bacteria on contact [399]or have sustained antimicrobial activity [400] should be further evaluated.

Several investigators have recognized heavy microbial contamination of wet mops and cleaning cloths and the potential for spread of such contamination [68, 401]. They have shown that wiping hard surfaces with contaminated cloths can contaminate hands, equipment, and other surfaces [68, 402]. Data

have been published that can be used to formulate effective policies for decontamination and maintenance of reusable cleaning cloths. For example, heat was the most reliable treatment of cleaning cloths as a detergent washing followed by drying at 80°C for 2 hours produced elimination of contamination. However, the dry heating process might be a fire hazard if the mop head contains petroleum-based products or lint builds up within the equipment or vent hose (American Health Care Association, personal communication, March 2003). Alternatively, immersing the cloth in hypochlorite (4,000 ppm) for 2 minutes produced no detectable surviving organisms in 10 of 13 cloths [403]. If reusable cleaning cloths or mops are used, they should be decontaminated regularly to prevent surface contamination during cleaning with subsequent transfer of organisms from these surfaces to patients or equipment by the hands of health-care workers. Some hospitals have begun using a new mopping technique involving microfiber materials to clean floors. Microfibers are densely constructed, polyester and polyamide (nylon) fibers, that are approximately 1/16 the thickness of a human hair. The positively charged microfibers attract dust (which has a negative charge) and are more absorbent than a conventional, cotton-loop mop. Microfiber materials also can be wet with disinfectants, such as quaternary ammonium compounds. In one study, the microfiber system tested demonstrated superior microbial removal compared with conventional string mops when used with a detergent cleaner (94% vs 68%). The use of a disinfectant did not improve the microbial elimination demonstrated by the microfiber system (95% vs 94%). However, use of disinfectant significantly improved microbial removal when a conventional string mop was used (95% vs 68%)(WA Rutala, unpublished data, August 2006). The microfiber system also prevents the possibility of transferring microbes from room to room because a new microfiber pad is used in each room.

Contact Times for Surface Disinfectants

An important issue concerning use of disinfectants for noncritical surfaces in health-care settings is that the contact time specified on the label of the product is often too long to be practically followed. The labels of most products registered by EPA for use against HBV, HIV, or *M. tuberculosis* specify a contact time of 10 minutes. Such a long contact time is not practical for disinfection of environmental surfaces in a health-care setting because most health-care facilities apply a disinfectant and allow it to dry (~1 minute). Multiple scientific papers have demonstrated significant microbial reduction with contact times of 30 to 60 seconds[46-56, 58-64]. In addition, EPA will approve a shortened contact time for any product for which the manufacturers will submit confirmatory efficacy data.

Currently, some EPA-registered disinfectants have contact times of one to three minutes. By law, users must follow all applicable label instructions for EPA-registered products. Ideally, product users should consider and use products that have the shortened contact time. However, disinfectant manufacturers also need to obtain EPA approval for shortened contact times so these products will be used correctly and effectively in the health-care environment.

Air Disinfection

Disinfectant spray-fog techniques for antimicrobial control in hospital rooms has been used. This technique of spraying of disinfectants is an unsatisfactory method of decontaminating air and surfaces and is not recommended for general infection control in routine patient-care areas[386]. Disinfectant fogging is rarely, if ever, used in U.S. healthcare facilities for air and surface disinfection in patient-care areas. Methods (e.g., filtration, ultraviolet germicidal irradiation, chlorine dioxide) to reduce air contamination in the healthcare setting are discussed in another guideline [23].

Microbial Contamination of Disinfectants

Contaminated disinfectants and antiseptics have been occasional vehicles of health-care infections and pseudoepidemics for more than 50 years. Published reports describing contaminated disinfectants and antiseptic solutions leading to health-care-associated infections have been summarized

[404]. Since this summary additional reports have been published [405-408]. An examination of reports of disinfectants contaminated with microorganisms revealed noteworthy observations. Perhaps most importantly, high-level disinfectants/liquid chemical sterilants have not been associated with outbreaks due to intrinsic or extrinsic contamination.Members of the genus *Pseudomonas* (e.g., *P. aeruginosa*) are the most frequent isolates from contaminated disinfectants—recovered from 80% of contaminated products. Their ability to remain viable or grow in use-dilutions of disinfectants is unparalleled. This survival advantage for *Pseudomonas* results presumably from their nutritional versatility, their unique outer membrane that constitutes an effective barrier to the passage of germicides, and/or efflux systems [409]. Although the concentrated solutions of the disinfectants have not been demonstrated to be contaminated at the point of manufacture, an undiluted phenolic can be contaminated by a *Pseudomonas* sp. during use [410]. In most of the reports that describe illness associated with contaminated disinfectants, the product was used to disinfect patient-care equipment, such as cystoscopes, cardiac catheters, and thermometers. Germicides used as disinfectants that were reported to have been contaminated include chlorhexidine, quaternary ammonium compounds, phenolics, and pine oil.

The following control measures should be instituted to reduce the frequency of bacterial growth in disinfectants and the threat of serious healthcare–associated infections from the use of such contaminated products [404]. First, some disinfectants should not be diluted; those that are diluted must be prepared correctly to achieve the manufacturers' recommended use-dilution. Second, infection-control professionals must learn from the literature what inappropriate activities result in extrinsic contamination (i.e., at the point of use) of germicides and train users to prevent recurrence. Common sources of extrinsic contamination of germicides in the reviewed literature are the water to make working dilutions, contaminated containers, and general contamination of the hospital areas where the germicides are prepared and/or used. Third, stock solutions of germicides must be stored as indicated on the product label. EPA verifies manufacturers' efficacy claims against microorganisms. These measures should provide assurance that products meeting the EPA registration requirements can achieve a certain level of antimicrobial activity when used as directed.

FACTORS AFFECTING THE EFFICACY OF DISINFECTION AND STERILIZATION

The activity of germicides against microorganisms depends on a number of factors, some of which are intrinsic qualities of the organism, others of which are the chemical and external physical environment. Awareness of these factors should lead to better use of disinfection and sterilization processes and will be briefly reviewed. More extensive consideration of these and other factors is available elsewhere [13, 14, 16, 411-413].

Number and Location of Microorganisms

All other conditions remaining constant, the larger the number of microbes, the more time a germicide needs to destroy all of them. Spaulding illustrated this relation when he employed identical test conditions and demonstrated that it took 30 minutes to kill 10 *B. atrophaeus* (formerly *Bacillus subtilis*) spores but 3 hours to kill 100,000 *Bacillus atrophaeus* spores. This reinforces the need for scrupulous cleaning of medical instruments before disinfection and sterilization. Reducing the number of microorganisms that must be inactivated through meticulous cleaning, increases the margin of safety when the germicide is used according to the labeling and shortens the exposure time required to kill the entire microbial load. Researchers also have shown that aggregated or clumped cells are more difficult to inactivate than monodispersed cells [414].

The location of microorganisms also must be considered when factors affecting the efficacy of germicides are assessed. Medical instruments with multiple pieces must be disassembled and equipment such as endoscopes that have crevices, joints, and channels are more difficult to disinfect than are flat-surface equipment because penetration of the disinfectant of all parts of the equipment is more difficult. Only surfaces that directly contact the germicide will be disinfected, so there must be no air pockets and the equipment must be completely immersed for the entire exposure period. Manufacturers should be encouraged to produce equipment engineered for ease of cleaning and disinfection.

Innate Resistance of Microorganisms

Microorganisms vary greatly in their resistance to chemical germicides and sterilization processes (Figure 1) [342] Intrinsic resistance mechanisms in microorganisms to disinfectants vary. For example, spores are resistant to disinfectants because the spore coat and cortex act as a barrier, mycobacteria have a waxy cell wall that prevents disinfectant entry, and gram-negative bacteria possess an outer membrane that acts as a barrier to the uptake of disinfectants [341, 343-345]. Implicit in all disinfection strategies is the consideration that the most resistant microbial subpopulation controls the sterilization or disinfection time. That is, to destroy the most resistant types of microorganisms (i.e., bacterial spores), the user needs to employ exposure times and a concentration of germicide needed to achieve complete destruction. Except for prions, bacterial spores possess the highest innate resistance to chemical germicides, followed by coccidia (e.g., *Cryptosporidium*), mycobacteria (e.g., *M. tuberculosis*), nonlipid or small viruses (e.g., poliovirus, and coxsackievirus), fungi (e.g., *Aspergillus,* and *Candida*), vegetative bacteria (e.g., *Staphylococcus,* and *Pseudomonas*) and lipid or medium-size viruses (e.g., herpes, and HIV). The germicidal resistance exhibited by the gram-positive and gram-negative bacteria is similar with some exceptions (e.g., *P. aeruginosa* which shows greater resistance to some disinfectants) [369, 415, 416]. *P. aeruginosa* also is significantly more resistant to a variety of disinfectants in its “naturally occurring” state than are cells subcultured on laboratory media [415, 417]. *Rickettsiae*, *Chlamydiae*, and mycoplasma cannot be placed in this scale of relative resistance because information about the efficacy of germicides against these agents is limited [418]. Because these microorganisms contain lipid and are similar in structure and composition to other bacteria, they can be predicted to be inactivated by the same germicides that destroy lipid viruses and vegetative bacteria. A known exception to this supposition is *Coxiella burnetti*, which has demonstrated resistance to disinfectants [419].

Concentration and Potency of Disinfectants

With other variables constant, and with one exception (iodophors), the more concentrated the

disinfectant, the greater its efficacy and the shorter the time necessary to achieve microbial kill. Generally not recognized, however, is that all disinfectants are not similarly affected by concentration adjustments. For example, quaternary ammonium compounds and phenol have a concentration exponent of 1 and 6, respectively; thus, halving the concentration of a quaternary ammonium compound requires doubling its disinfecting time, but halving the concentration of a phenol solution requires a 64-fold (i.e., 2^6) increase in its disinfecting time [365, 413, 420].

Considering the length of the disinfection time, which depends on the potency of the germicide, also is important. This was illustrated by Spaulding who demonstrated using the mucin-loop test that 70% isopropyl alcohol destroyed 10^4 *M. tuberculosis* in 5 minutes, whereas a simultaneous test with 3% phenolic required 2–3 hours to achieve the same level of microbial kill [14].

Physical and Chemical Factors

Several physical and chemical factors also influence disinfectant procedures: temperature, pH, relative humidity, and water hardness. For example, the activity of most disinfectants increases as the temperature increases, but some exceptions exist. Furthermore, too great an increase in temperature causes the disinfectant to degrade and weakens its germicidal activity and thus might produce a potential health hazard.

An increase in pH improves the antimicrobial activity of some disinfectants (e.g., glutaraldehyde, quaternary ammonium compounds) but decreases the antimicrobial activity of others (e.g., phenols, hypochlorites, and iodine). The pH influences the antimicrobial activity by altering the disinfectant molecule or the cell surface [413].

Relative humidity is the single most important factor influencing the activity of gaseous disinfectants/sterilants, such as EtO, chlorine dioxide, and formaldehyde.

Water hardness (i.e., high concentration of divalent cations) reduces the rate of kill of certain disinfectants because divalent cations (e.g., magnesium, calcium) in the hard water interact with the disinfectant to form insoluble precipitates [13, 421].

Organic and Inorganic Matter

Organic matter in the form of serum, blood, pus, or fecal or lubricant material can interfere with the antimicrobial activity of disinfectants in at least two ways. Most commonly, interference occurs by a chemical reaction between the germicide and the organic matter resulting in a complex that is less germicidal or nongermicidal, leaving less of the active germicide available for attacking microorganisms. Chlorine and iodine disinfectants, in particular, are prone to such interaction. Alternatively, organic material can protect microorganisms from attack by acting as a physical barrier [422, 423].

The effects of inorganic contaminants on the sterilization process were studied during the 1950s and 1960s [424, 425]. These and other studies show the protection by inorganic contaminants of microorganisms to all sterilization processes results from occlusion in salt crystals [426, 427]. This further emphasizes the importance of meticulous cleaning of medical devices before any sterilization or disinfection procedure because both organic and inorganic soils are easily removed by washing [426].

Duration of Exposure

Items must be exposed to the germicide for the appropriate minimum contact time. Multiple investigators have demonstrated the effectiveness of low-level disinfectants against vegetative bacteria (e.g., *Listeria*, *E. coli*, *Salmonella*, VRE, MRSA), yeasts (e.g., *Candida*), mycobacteria (e.g., *M. tuberculosis*), and viruses (e.g., poliovirus) at exposure times of 30–60 seconds [46-64]. By law, all applicable label instructions on EPA-registered products must be followed. If the user selects exposure conditions that differ from those on the EPA-registered product label, the user assumes liability for any injuries resulting from off-label use and is potentially subject to enforcement action under the Federal Insecticide, Fungicide, and Rodenticide Act (FIFRA)

All lumens and channels of endoscopic instruments must contact the disinfectant. Air pockets interfere with the disinfection process, and items that float on the disinfectant will not be disinfected. The disinfectant must be introduced reliably into the internal channels of the device. The exact times for disinfecting medical items are somewhat elusive because of the effect of the aforementioned factors on disinfection efficacy. Certain contact times have proved reliable (Table 1), but, in general, longer contact times are more effective than shorter contact times.

Biofilms

Microorganisms may be protected from disinfectants by production of thick masses of cells [428] and extracellular materials, or biofilms [429-435]. Biofilms are microbial communities that are tightly attached to surfaces and cannot be easily removed. Once these masses form, microbes within them can be resistant to disinfectants by multiple mechanisms, including physical characteristics of older biofilms, genotypic variation of the bacteria, microbial production of neutralizing enzymes, and physiologic gradients within the biofilm (e.g., pH). Bacteria within biofilms are up to 1,000 times more resistant to antimicrobials than are the same bacteria in suspension [436]. Although new decontamination methods [437] are being investigated for removing biofilms, chlorine and monochloramines can effectively inactivate biofilm bacteria [431, 438]. Investigators have hypothesized that the glycocalyx-like cellular masses on the interior walls of polyvinyl chloride pipe would protect embedded organisms from some disinfectants and be a reservoir for continuous contamination [429, 430, 439]. Biofilms have been found in whirlpools [440], dental unit waterlines[441], and numerous medical devices (e.g., contact lenses, pacemakers, hemodialysis systems, urinary catheters, central venous catheters, endoscopes) [434, 436, 438, 442]. Their presence can have serious implications for immunocompromised patients and patients who have indwelling medical devices. Some enzymes [436, 443, 444] and detergents [436] can degrade biofilms or reduce numbers of viable bacteria within a biofilm, but no products are EPA-registered or FDA-cleared for this purpose.

CLEANING

Cleaning is the removal of foreign material (e.g., soil, and organic material) from objects and is normally accomplished using water with detergents or enzymatic products. Thorough cleaning is required before high-level disinfection and sterilization because inorganic and organic materials that remain on the surfaces of instruments interfere with the effectiveness of these processes. Also, if soiled materials dry or bake onto the instruments, the removal process becomes more difficult and the disinfection or sterilization process less effective or ineffective. Surgical instruments should be presoaked or rinsed to prevent drying of blood and to soften or remove blood from the instruments.

Cleaning is done manually in use areas without mechanical units (e.g., ultrasonic cleaners or washer-disinfectors) or for fragile or difficult-to-clean instruments. With manual cleaning, the two essential components are friction and fluidics. Friction (e.g., rubbing/scrubbing the soiled area with a brush) is an old and dependable method. Fluidics (i.e., fluids under pressure) is used to remove soil and debris from internal channels after brushing and when the design does not allow passage of a brush through a channel [445]. When a washer-disinfector is used, care should be taken in loading instruments: hinged instruments should be opened fully to allow adequate contact with the detergent solution; stacking of instruments in washers should be avoided; and instruments should be disassembled as much as possible.

The most common types of mechanical or automatic cleaners are ultrasonic cleaners, washer-decontaminators, washer-disinfectors, and washer-sterilizers. Ultrasonic cleaning removes soil by cavitation and implosion in which waves of acoustic energy are propagated in aqueous solutions to disrupt the bonds that hold particulate matter to surfaces. Bacterial contamination can be present in used ultrasonic cleaning solutions (and other used detergent solutions) because these solutions generally do not make antibacterial label claims [446]. Even though ultrasound alone does not significantly inactivate bacteria, sonication can act synergistically to increase the cidal efficacy of a disinfectant [447]. Users of ultrasonic cleaners should be aware that the cleaning fluid could result in endotoxin contamination of surgical instruments, which could cause severe inflammatory reactions [448]. Washer-sterilizers are modified steam sterilizers that clean by filling the chamber with water and detergent through which steam passes to provide agitation. Instruments are subsequently rinsed and subjected to a short steam-sterilization cycle. Another washer-sterilizer employs rotating spray arms for a wash cycle followed by a steam sterilization cycle at 285°F [449, 450]. Washer-decontaminators/disinfectors act like a dishwasher that uses a combination of water circulation and detergents to remove soil. These units sometimes have a cycle that subjects the instruments to a heat process (e.g., 93°C for 10 minutes) [451]. Washer-disinfectors are generally computer-controlled units for cleaning, disinfecting, and drying solid and hollow surgical and medical equipment. In one study, cleaning (measured as 5–6 $\log_{10}$ reduction) was achieved on surfaces that had adequate contact with the water flow in the machine [452]. Detailed information about cleaning and preparing supplies for terminal sterilization is provided by professional organizations [453, 454] and books [455]. Studies have shown that manual and mechanical cleaning of endoscopes achieves approximately a 4-$\log_{10}$ reduction of contaminating organisms [83, 104, 456, 457]. Thus, cleaning alone effectively reduces the number of microorganisms on contaminated equipment. In a quantitative analysis of residual protein contamination of reprocessed surgical instruments, median levels of residual protein contamination per instrument for five trays were 267, 260, 163, 456, and 756 µg [458]. In another study, the median amount of protein from reprocessed surgical instruments from different hospitals ranged from 8 µg to 91 µg [459]. When manual methods were compared with automated methods for cleaning reusable accessory devices used for minimally invasive surgical procedures, the automated method was more efficient for cleaning biopsy forceps and ported and nonported laparoscopic devices and achieved a >99% reduction in soil parameters (i.e., protein, carbohydrate, hemoglobin) in the ported and nonported laparoscopic devices [460, 461]

For instrument cleaning, a neutral or near-neutral pH detergent solution commonly is used because such solutions generally provide the best material compatibility profile and good soil removal.

Enzymes, usually proteases, sometimes are added to neutral pH solutions to assist in removing organic material. Enzymes in these formulations attack proteins that make up a large portion of common soil (e.g., blood, pus). Cleaning solutions also can contain lipases (enzymes active on fats) and amylases (enzymes active on starches). Enzymatic cleaners are not disinfectants, and proteinaceous enzymes can be inactivated by germicides. As with all chemicals, enzymes must be rinsed from the equipment or adverse reactions (e.g., fever, residual amounts of high-level disinfectants, proteinaceous residue) could result [462, 463]. Enzyme solutions should be used in accordance with manufacturer's instructions, which include proper dilution of the enzymatic detergent and contact with equipment for the amount of time specified on the label [463]. Detergent enzymes can result in asthma or other allergic effects in users. Neutral pH detergent solutions that contain enzymes are compatible with metals and other materials used in medical instruments and are the best choice for cleaning delicate medical instruments, especially flexible endoscopes [457]. Alkaline-based cleaning agents are used for processing medical devices because they efficiently dissolve protein and fat residues [464]; however, they can be corrosive [457]. Some data demonstrate that enzymatic cleaners are more effective than neutral detergents [465, 466] in removing microorganisms from surfaces but two more recent studies found no difference in cleaning efficiency between enzymatic and alkaline-based cleaners [443, 464]. Another study found no significant difference between enzymatic and non-enzymatic cleaners in terms of microbial cleaning efficacy [467]. A new non-enzyme, hydrogen peroxide-based formulation (not FDA-cleared) was as effective as enzymatic cleaners in removing protein, blood, carbohydrate, and endotoxin from surface test carriers[468] In addition, this product effected a 5-$\log_{10}$ reduction in microbial loads with a 3-minute exposure at room temperature [468].

Although the effectiveness of high-level disinfection and sterilization mandates effective cleaning, no "real-time" tests exist that can be employed in a clinical setting to verify cleaning. If such tests were commercially available they could be used to ensure an adequate level of cleaning [469-472].). The only way to ensure adequate cleaning is to conduct a reprocessing verification test (e.g., microbiologic sampling), but this is not routinely recommended [473]. Validation of the cleaning processes in a laboratory-testing program is possible by microorganism detection, chemical detection for organic contaminants, radionuclide tagging, and chemical detection for specific ions [426, 471]. During the past few years, data have been published describing use of an artificial soil, protein, endotoxin, X-ray contrast medium, or blood to verify the manual or automated cleaning process [169, 452, 474-478] and adenosine triphosphate bioluminescence and microbiologic sampling to evaluate the effectiveness of environmental surface cleaning[170, 479]. At a minimum, all instruments should be individually inspected and be visibly clean.

DISINFECTION

Many disinfectants are used alone or in combinations (e.g., hydrogen peroxide and peracetic acid) in the health-care setting. These include alcohols, chlorine and chlorine compounds, formaldehyde, glutaraldehyde, *ortho*-phthalaldehyde, hydrogen peroxide, iodophors, peracetic acid, phenolics, and quaternary ammonium compounds. Commercial formulations based on these chemicals are considered unique products and must be registered with EPA or cleared by FDA. In most instances, a given product is designed for a specific purpose and is to be used in a certain manner. Therefore, users should read labels carefully to ensure the correct product is selected for the intended use and applied efficiently.

Disinfectants are not interchangeable, and incorrect concentrations and inappropriate disinfectants can result in excessive costs. Because occupational diseases among cleaning personnel have been associated with use of several disinfectants (e.g., formaldehyde, glutaraldehyde, and chlorine), precautions (e.g., gloves and proper ventilation) should be used to minimize exposure [318, 480, 481]. Asthma and reactive airway disease can occur in sensitized persons exposed to any airborne chemical, including germicides. Clinically important asthma can occur at levels below ceiling levels regulated by OSHA or recommended by NIOSH. The preferred method of control is elimination of the chemical (through engineering controls or substitution) or relocation of the worker.

The following overview of the performance characteristics of each provides users with sufficient information to select an appropriate disinfectant for any item and use it in the most efficient way.

Chemical Disinfectants

Alcohol

Overview. In the healthcare setting, "alcohol" refers to two water-soluble chemical compounds—ethyl alcohol and isopropyl alcohol—that have generally underrated germicidal characteristics [482]. FDA has not cleared any liquid chemical sterilant or high-level disinfectant with alcohol as the main active ingredient. These alcohols are rapidly bactericidal rather than bacteriostatic against vegetative forms of bacteria; they also are tuberculocidal, fungicidal, and virucidal but do not destroy bacterial spores. Their cidal activity drops sharply when diluted below 50% concentration, and the optimum bactericidal concentration is 60%–90% solutions in water (volume/volume) [483, 484].

Mode of Action. The most feasible explanation for the antimicrobial action of alcohol is denaturation of proteins. This mechanism is supported by the observation that absolute ethyl alcohol, a dehydrating agent, is less bactericidal than mixtures of alcohol and water because proteins are denatured more quickly in the presence of water [484, 485]. Protein denaturation also is consistent with observations that alcohol destroys the dehydrogenases of *Escherichia coli* [486], and that ethyl alcohol increases the lag phase of *Enterobacter aerogenes* [487] and that the lag phase effect could be reversed by adding certain amino acids. The bacteriostatic action was believed caused by inhibition of the production of metabolites essential for rapid cell division.

Microbicidal Activity. Methyl alcohol (methanol) has the weakest bactericidal action of the alcohols and thus seldom is used in healthcare [488]. The bactericidal activity of various concentrations of ethyl alcohol (ethanol) was examined against a variety of microorganisms in exposure periods ranging from 10 seconds to 1 hour [483]. *Pseudomonas aeruginosa* was killed in 10 seconds by all concentrations of ethanol from 30% to 100% (v/v), and *Serratia marcescens, E, coli* and *Salmonella typhosa* were killed in 10 seconds by all concentrations of ethanol from 40% to 100%. The gram-positive organisms *Staphylococcus aureus* and *Streptococcus pyogenes* were slightly more resistant, being killed in 10 seconds by ethyl alcohol concentrations of 60%–95%. Isopropyl alcohol (isopropanol) was slightly more bactericidal than ethyl alcohol for *E. coli* and *S. aureus* [489].

Ethyl alcohol, at concentrations of 60%–80%, is a potent virucidal agent inactivating all of the lipophilic viruses (e.g., herpes, vaccinia, and influenza virus) and many hydrophilic viruses (e.g.,

adenovirus, enterovirus, rhinovirus, and rotaviruses but not hepatitis A virus (HAV) [58] or poliovirus) [49]. Isopropyl alcohol is not active against the nonlipid enteroviruses but is fully active against the lipid viruses [72]. Studies also have demonstrated the ability of ethyl and isopropyl alcohol to inactivate the hepatitis B virus(HBV) [224, 225] and the herpes virus, [490] and ethyl alcohol to inactivate human immunodeficiency virus (HIV) [227], rotavirus, echovirus, and astrovirus [491].

In tests of the effect of ethyl alcohol against *M. tuberculosis*, 95% ethanol killed the tubercle bacilli in sputum or water suspension within 15 seconds [492]. In 1964, Spaulding stated that alcohols were the germicide of choice for tuberculocidal activity, and they should be the standard by which all other tuberculocides are compared. For example, he compared the tuberculocidal activity of iodophor (450 ppm), a substituted phenol (3%), and isopropanol (70%/volume) using the mucin-loop test (10^6 *M. tuberculosis* per loop) and determined the contact times needed for complete destruction were 120–180 minutes, 45–60 minutes, and 5 minutes, respectively. The mucin-loop test is a severe test developed to produce long survival times. Thus, these figures should not be extrapolated to the exposure times needed when these germicides are used on medical or surgical material [482].

Ethyl alcohol (70%) was the most effective concentration for killing the tissue phase of *Cryptococcus neoformans*, *Blastomyces dermatitidis*, *Coccidioides immitis*, and *Histoplasma capsulatum* and the culture phases of the latter three organisms aerosolized onto various surfaces. The culture phase was more resistant to the action of ethyl alcohol and required about 20 minutes to disinfect the contaminated surface, compared with <1 minute for the tissue phase [493, 494].

Isopropyl alcohol (20%) is effective in killing the cysts of *Acanthamoeba culbertsoni* (*560*) as are chlorhexidine, hydrogen peroxide, and thimerosal [496].

Uses. Alcohols are not recommended for sterilizing medical and surgical materials principally because they lack sporicidal action and they cannot penetrate protein-rich materials. Fatal postoperative wound infections with *Clostridium* have occurred when alcohols were used to sterilize surgical instruments contaminated with bacterial spores [497]. Alcohols have been used effectively to disinfect oral and rectal thermometers[498, 499], hospital pagers [500], scissors [501], and stethoscopes [502]. Alcohols have been used to disinfect fiberoptic endoscopes [503, 504] but failure of this disinfectant have lead to infection [280, 505]. Alcohol towelettes have been used for years to disinfect small surfaces such as rubber stoppers of multiple-dose medication vials or vaccine bottles. Furthermore, alcohol occasionally is used to disinfect external surfaces of equipment (e.g., stethoscopes, ventilators, manual ventilation bags) [506], CPR manikins [507], ultrasound instruments [508] or medication preparation areas. Two studies demonstrated the effectiveness of 70% isopropyl alcohol to disinfect reusable transducer heads in a controlled environment [509, 510]. In contrast, three bloodstream infection outbreaks have been described when alcohol was used to disinfect transducer heads in an intensive-care setting [511].

The documented shortcomings of alcohols on equipment are that they damage the shellac mountings of lensed instruments, tend to swell and harden rubber and certain plastic tubing after prolonged and repeated use, bleach rubber and plastic tiles [482] and damage tonometer tips (by deterioration of the glue) after the equivalent of 1 working year of routine use [512]. Tonometer biprisms soaked in alcohol for 4 days developed rough front surfaces that potentially could cause corneal damage; this appeared to be caused by weakening of the cementing substances used to fabricate the biprisms [513]. Corneal opacification has been reported when tonometer tips were swabbed with alcohol immediately before measurement of intraocular pressure [514]. Alcohols are flammable and consequently must be stored in a cool, well-ventilated area. They also evaporate rapidly, making extended exposure time difficult to achieve unless the items are immersed.

Chlorine and Chlorine Compounds

Overview. Hypochlorites, the most widely used of the chlorine disinfectants, are available as liquid (e.g., sodium hypochlorite) or solid (e.g., calcium hypochlorite). The most prevalent chlorine

products in the United States are aqueous solutions of 5.25%–6.15% sodium hypochlorite (see glossary), usually called household bleach. They have a broad spectrum of antimicrobial activity, do not leave toxic residues, are unaffected by water hardness, are inexpensive and fast acting [328], remove dried or fixed organisms and biofilms from surfaces[465], and have a low incidence of serious toxicity [515-517]. Sodium hypochlorite at the concentration used in household bleach (5.25-6.15%) can produce ocular irritation or oropharyngeal, esophageal, and gastric burns [318, 518-522]. Other disadvantages of hypochlorites include corrosiveness to metals in high concentrations (>500 ppm), inactivation by organic matter, discoloring or "bleaching" of fabrics, release of toxic chlorine gas when mixed with ammonia or acid (e.g., household cleaning agents) [523-525], and relative stability [327]. The microbicidal activity of chlorine is attributed largely to undissociated hypochlorous acid (HOCl). The dissociation of HOCl to the less microbicidal form (hypochlorite ion OCl^-) depends on pH. The disinfecting efficacy of chlorine decreases with an increase in pH that parallels the conversion of undissociated HOCl to OCl^- [329, 526]. A potential hazard is production of the carcinogen bis(chloromethyl) ether when hypochlorite solutions contact formaldehyde [527] and the production of the animal carcinogen trihalomethane when hot water is hyperchlorinated [528]. After reviewing environmental fate and ecologic data, EPA has determined the currently registered uses of hypochlorites will not result in unreasonable adverse effects to the environment [529].

Alternative compounds that release chlorine and are used in the health-care setting include demand-release chlorine dioxide, sodium dichloroisocyanurate, and chloramine-T. The advantage of these compounds over the hypochlorites is that they retain chlorine longer and so exert a more prolonged bactericidal effect. Sodium dichloroisocyanurate tablets are stable, and for two reasons, the microbicidal activity of solutions prepared from sodium dichloroisocyanurate tablets might be greater than that of sodium hypochlorite solutions containing the same total available chlorine. First, with sodium dichloroisocyanurate, only 50% of the total available chlorine is free (HOCl and OCl^-), whereas the remainder is combined (monochloroisocyanurate or dichloroisocyanurate), and as free available chlorine is used up, the latter is released to restore the equilibrium. Second, solutions of sodium dichloroisocyanurate are acidic, whereas sodium hypochlorite solutions are alkaline, and the more microbicidal type of chlorine (HOCl) is believed to predominate [530-533]. Chlorine dioxide-based disinfectants are prepared fresh as required by mixing the two components (base solution [citric acid with preservatives and corrosion inhibitors] and the activator solution [sodium chlorite]). In vitro suspension tests showed that solutions containing about 140 ppm chlorine dioxide achieved a reduction factor exceeding 10^6 of *S. aureus* in 1 minute and of *Bacillus atrophaeus* spores in 2.5 minutes in the presence of 3 g/L bovine albumin. The potential for damaging equipment requires consideration because long-term use can damage the outer plastic coat of the insertion tube [534]. In another study, chlorine dioxide solutions at either 600 ppm or 30 ppm killed *Mycobacterium avium-intracellulare* within 60 seconds after contact but contamination by organic material significantly affected the microbicidal properties[535].

The microbicidal activity of a new disinfectant, "superoxidized water," has been examined The concept of electrolyzing saline to create a disinfectant or antiseptics is appealing because the basic materials of saline and electricity are inexpensive and the end product (i.e., water) does not damage the environment. The main products of this water are hypochlorous acid (e.g., at a concentration of about 144 mg/L) and chlorine. As with any germicide, the antimicrobial activity of superoxidized water is strongly affected by the concentration of the active ingredient (available free chlorine) [536]. One manufacturer generates the disinfectant at the point of use by passing a saline solution over coated titanium electrodes at 9 amps. The product generated has a pH of 5.0–6.5 and an oxidation-reduction potential (redox) of >950 mV. Although superoxidized water is intended to be generated fresh at the point of use, when tested under clean conditions the disinfectant was effective within 5 minutes when 48 hours old [537]. Unfortunately, the equipment required to produce the product can be expensive because parameters such as pH, current, and redox potential must be closely monitored. The solution is nontoxic to biologic tissues. Although the United Kingdom manufacturer claims the solution is noncorrosive and nondamaging to endoscopes and processing equipment, one flexible endoscope manufacturer (Olympus Key-Med, United Kingdom) has voided the warranty on the endoscopes if superoxidized water is used to disinfect them [538]. As with any germicide formulation, the user should check with the device manufacturer for

compatibility with the germicide. Additional studies are needed to determine whether this solution could be used as an alternative to other disinfectants or antiseptics for hand washing, skin antisepsis, room cleaning, or equipment disinfection (e.g., endoscopes, dialyzers) [400, 539, 540]. In October 2002, the FDA cleared superoxidized water as a high-level disinfectant (FDA, personal communication, September 18, 2002).

Mode of Action. The exact mechanism by which free chlorine destroys microorganisms has not been elucidated. Inactivation by chlorine can result from a number of factors: oxidation of sulfhydryl enzymes and amino acids; ring chlorination of amino acids; loss of intracellular contents; decreased uptake of nutrients; inhibition of protein synthesis; decreased oxygen uptake; oxidation of respiratory components; decreased adenosine triphosphate production; breaks in DNA; and depressed DNA synthesis [329, 347]. The actual microbicidal mechanism of chlorine might involve a combination of these factors or the effect of chlorine on critical sites [347].

Microbicidal Activity. Low concentrations of free available chlorine (e.g., HOCl, OCl^-, and elemental chlorine-Cl_2) have a biocidal effect on mycoplasma (25 ppm) and vegetative bacteria (<5 ppm) in seconds in the absence of an organic load [329, 418]. Higher concentrations (1,000 ppm) of chlorine are required to kill *M. tuberculosis* using the Association of Official Analytical Chemists (AOAC) tuberculocidal test [73]. A concentration of 100 ppm will kill ≥99.9% of *B. atrophaeus* spores within 5 minutes [541, 542] and destroy mycotic agents in <1 hour [329]. Acidified bleach and regular bleach (5,000 ppm chlorine) can inactivate 10^6 *Clostridium difficile* spores in ≤10 minutes [262]. One study reported that 25 different viruses were inactivated in 10 minutes with 200 ppm available chlorine [72]. Several studies have demonstrated the effectiveness of diluted sodium hypochlorite and other disinfectants to inactivate HIV [61]. Chlorine (500 ppm) showed inhibition of *Candida* after 30 seconds of exposure [54]. In experiments using the AOAC Use-Dilution Method, 100 ppm of free chlorine killed 10^6–10^7 *S. aureus*, *Salmonella choleraesuis*, and *P. aeruginosa* in <10 minutes [327]. Because household bleach contains 5.25%–6.15% sodium hypochlorite, or 52,500–61,500 ppm available chlorine, a 1:1,000 dilution provides about 53–62 ppm available chlorine, and a 1:10 dilution of household bleach provides about 5250–6150 ppm.

Data are available for chlorine dioxide that support manufacturers' bactericidal, fungicidal, sporicidal, tuberculocidal, and virucidal label claims [543-546]. A chlorine dioxide generator has been shown effective for decontaminating flexible endoscopes [534] but it is not currently FDA-cleared for use as a high-level disinfectant [85]. Chlorine dioxide can be produced by mixing solutions, such as a solution of chlorine with a solution of sodium chlorite [329]. In 1986, a chlorine dioxide product was voluntarily removed from the market when its use caused leakage of cellulose-based dialyzer membranes, which allowed bacteria to migrate from the dialysis fluid side of the dialyzer to the blood side [547].

Sodium dichloroisocyanurate at 2,500 ppm available chlorine is effective against bacteria in the presence of up to 20% plasma, compared with 10% plasma for sodium hypochlorite at 2,500 ppm [548].

"Superoxidized water" has been tested against bacteria, mycobacteria, viruses, fungi, and spores [537, 539, 549]. Freshly generated superoxidized water is rapidly effective (<2 minutes) in achieving a 5-$\log_{10}$ reduction of pathogenic microorganisms (i.e., *M. tuberculosis*, *M. chelonae*, poliovirus, HIV, multidrug-resistant *S. aureus*, *E. coli, Candida albicans*, *Enterococcus faecalis, P. aeruginosa*) in the absence of organic loading. However, the biocidal activity of this disinfectant decreased substantially in the presence of organic material (e.g., 5% horse serum) [537, 549, 550]. No bacteria or viruses were detected on artificially contaminated endoscopes after a 5-minute exposure to superoxidized water [551] and HBV-DNA was not detected from any endoscope experimentally contaminated with HBV-positive mixed sera after a disinfectant exposure time of 7 minutes[552].

Uses. Hypochlorites are widely used in healthcare facilities in a variety of settings. [328] Inorganic chlorine solution is used for disinfecting tonometer heads [188] and for spot-disinfection of countertops and floors. A 1:10–1:100 dilution of 5.25%–6.15% sodium hypochlorite (i.e., household bleach) [22, 228, 553, 554] or

an EPA-registered tuberculocidal disinfectant [17]has been recommended for decontaminating blood spills. For small spills of blood (i.e., drops of blood) on noncritical surfaces, the area can be disinfected with a 1:100 dilution of 5.25%-6.15% sodium hypochlorite or an EPA-registered tuberculocidal disinfectant. Because hypochlorites and other germicides are substantially inactivated in the presence of blood [63, 548, 555, 556], large spills of blood require that the surface be cleaned before an EPA-registered disinfectant or a 1:10 (final concentration) solution of household bleach is applied [557]. If a sharps injury is possible, the surface initially should be decontaminated [69, 318], then cleaned and disinfected (1:10 final concentration) [63]. Extreme care always should be taken to prevent percutaneous injury. At least 500 ppm available chlorine for 10 minutes is recommended for decontaminating CPR training manikins [558]. Full-strength bleach has been recommended for self-disinfection of needles and syringes used for illicit-drug injection when needle-exchange programs are not available. The difference in the recommended concentrations of bleach reflects the difficulty of cleaning the interior of needles and syringes and the use of needles and syringes for parenteral injection [559]. Clinicians should not alter their use of chlorine on environmental surfaces on the basis of testing methodologies that do not simulate actual disinfection practices [560, 561]. Other uses in healthcare include as an irrigating agent in endodontic treatment [562] and as a disinfectant for manikins, laundry, dental appliances, hydrotherapy tanks [23, 41], regulated medical waste before disposal [328], and the water distribution system in hemodialysis centers and hemodialysis machines [563].

Chlorine long has been used as the disinfectant in water treatment. Hyperchlorination of a *Legionella*-contaminated hospital water system [23] resulted in a dramatic decrease (from 30% to 1.5%) in the isolation of *L. pneumophila* from water outlets and a cessation of healthcare-associated Legionnaires' disease in an affected unit [528, 564]. Water disinfection with monochloramine by municipal water-treatment plants substantially reduced the risk for healthcare–associated Legionnaires disease [565, 566]. Chlorine dioxide also has been used to control *Legionella* in a hospital water supply. [567] Chloramine T [568] and hypochlorites [41] have been used to disinfect hydrotherapy equipment.

Hypochlorite solutions in tap water at a pH >8 stored at room temperature (23°C) in closed, opaque plastic containers can lose up to 40%–50% of their free available chlorine level over 1 month. Thus, if a user wished to have a solution containing 500 ppm of available chlorine at day 30, he or she should prepare a solution containing 1,000 ppm of chlorine at time 0. Sodium hypochlorite solution does not decompose after 30 days when stored in a closed brown bottle [327].

The use of powders, composed of a mixture of a chlorine-releasing agent with highly absorbent resin, for disinfecting spills of body fluids has been evaluated by laboratory tests and hospital ward trials. The inclusion of acrylic resin particles in formulations markedly increases the volume of fluid that can be soaked up because the resin can absorb 200–300 times its own weight of fluid, depending on the fluid consistency. When experimental formulations containing 1%, 5%, and 10% available chlorine were evaluated by a standardized surface test, those containing 10% demonstrated bactericidal activity. One problem with chlorine-releasing granules is that they can generate chlorine fumes when applied to urine [569].

Formaldehyde

Overview. Formaldehyde is used as a disinfectant and sterilant in both its liquid and gaseous states. Liquid formaldehyde will be considered briefly in this section, and the gaseous form is reviewed elsewhere [570]. Formaldehyde is sold and used principally as a water-based solution called formalin, which is 37% formaldehyde by weight. The aqueous solution is a bactericide, tuberculocide, fungicide, virucide and sporicide [72, 82, 571-573]. OSHA indicated that formaldehyde should be handled in the workplace as a potential carcinogen and set an employee exposure standard for formaldehyde that limits an 8-hour time-weighted average exposure concentration of 0.75 ppm [574, 575]. The standard includes a second permissible exposure limit in the form of a short-term exposure limit (STEL) of 2 ppm that is the maximum exposure allowed during a 15-minute period [576]. Ingestion of formaldehyde can be fatal, and long-term exposure to low levels in the air or on the skin can cause asthma-like respiratory problems and skin irritation, such as dermatitis and itching. For these reasons, employees should have limited direct contact

with formaldehyde, and these considerations limit its role in sterilization and disinfection processes. Key provisions of the OSHA standard that protects workers from exposure to formaldehyde appear in Title 29 of the Code of Federal Regulations (CFR) Part 1910.1048 (and equivalent regulations in states with OSHA-approved state plans) [577].

Mode of Action. Formaldehyde inactivates microorganisms by alkylating the amino and sulfhydral groups of proteins and ring nitrogen atoms of purine bases [376].

Microbicidal Activity. Varying concentrations of aqueous formaldehyde solutions destroy a wide range of microorganisms. Inactivation of poliovirus in 10 minutes required an 8% concentration of formalin, but all other viruses tested were inactivated with 2% formalin [72]. Four percent formaldehyde is a tuberculocidal agent, inactivating 10^4 *M. tuberculosis* in 2 minutes [82], and 2.5% formaldehyde inactivated about 10^7 *Salmonella* Typhi in 10 minutes in the presence of organic matter [572]. The sporicidal action of formaldehyde was slower than that of glutaraldehyde in comparative tests with 4% aqueous formaldehyde and 2% glutaraldehyde against the spores of *B. anthracis* [82]. The formaldehyde solution required 2 hours of contact to achieve an inactivation factor of 10^4, whereas glutaraldehyde required only 15 minutes.

Uses. Although formaldehyde-alcohol is a chemical sterilant and formaldehyde is a high-level disinfectant, the health-care uses of formaldehyde are limited by its irritating fumes and its pungent odor even at very low levels (<1 ppm). For these reasons and others—such as its role as a suspected human carcinogen linked to nasal cancer and lung cancer [578], this germicide is excluded from Table 1. When it is used, , direct exposure to employees generally is limited; however, excessive exposures to formaldehyde have been documented for employees of renal transplant units [574, 579], and students in a gross anatomy laboratory [580]. Formaldehyde is used in the health-care setting to prepare viral vaccines (e.g., poliovirus and influenza); as an embalming agent; and to preserve anatomic specimens; and historically has been used to sterilize surgical instruments, especially when mixed with ethanol. A 1997 survey found that formaldehyde was used for reprocessing hemodialyzers by 34% of U.S. hemodialysis centers—a 60% decrease from 1983 [249, 581]. If used at room temperature, a concentration of 4% with a minimum exposure of 24 hours is required to disinfect disposable hemodialyzers reused on the same patient [582, 583]. Aqueous formaldehyde solutions (1%–2%) also have been used to disinfect the internal fluid pathways of dialysis machines [583]. To minimize a potential health hazard to dialysis patients, the dialysis equipment must be thoroughly rinsed and tested for residual formaldehyde before use.

Paraformaldehyde, a solid polymer of formaldehyde, can be vaporized by heat for the gaseous decontamination of laminar flow biologic safety cabinets when maintenance work or filter changes require access to the sealed portion of the cabinet.

Glutaraldehyde

Overview. Glutaraldehyde is a saturated dialdehyde that has gained wide acceptance as a high-level disinfectant and chemical sterilant [107]. Aqueous solutions of glutaraldehyde are acidic and generally in this state are not sporicidal. Only when the solution is “activated” (made alkaline) by use of alkalinating agents to pH 7.5–8.5 does the solution become sporicidal. Once activated, these solutions have a shelf-life of minimally 14 days because of the polymerization of the glutaraldehyde molecules at alkaline pH levels. This polymerization blocks the active sites (aldehyde groups) of the glutaraldehyde molecules that are responsible for its biocidal activity.

Novel glutaraldehyde formulations (e.g., glutaraldehyde-phenol-sodium phenate, potentiated acid glutaraldehyde, stabilized alkaline glutaraldehyde) produced in the past 30 years have overcome the problem of rapid loss of activity (e.g., use-life 28–30 days) while generally maintaining excellent microbicidal activity [584-588]. However, antimicrobial activity depends not only on age but also on use conditions, such as dilution and organic stress. Manufacturers' literature for these preparations suggests the neutral or alkaline glutaraldehydes possess microbicidal and anticorrosion properties superior to

those of acid glutaraldehydes, and a few published reports substantiate these claims [542, 589, 590]. However, two studies found no difference in the microbicidal activity of alkaline and acid glutaraldehydes [73, 591]. The use of glutaraldehyde-based solutions in health-care facilities is widespread because of their advantages, including excellent biocidal properties; activity in the presence of organic matter (20% bovine serum); and noncorrosive action to endoscopic equipment, thermometers, rubber, or plastic equipment (Tables 4 and 5).

Mode of Action. The biocidal activity of glutaraldehyde results from its alkylation of sulfhydryl, hydroxyl, carboxyl, and amino groups of microorganisms, which alters RNA, DNA, and protein synthesis. The mechanism of action of glutaraldehydes are reviewed extensively elsewhere [592, 593].

Microbicidal Activity. The in vitro inactivation of microorganisms by glutaraldehydes has been extensively investigated and reviewed [592, 593]. Several investigators showed that ≥2% aqueous solutions of glutaraldehyde, buffered to pH 7.5–8.5 with sodium bicarbonate effectively killed vegetative bacteria in <2 minutes; *M. tuberculosis*, fungi, and viruses in <10 minutes; and spores of *Bacillus* and *Clostridium* species in 3 hours [542, 592-597]. Spores of *C. difficile* are more rapidly killed by 2% glutaraldehyde than are spores of other species of *Clostridium* and *Bacillus* [79, 265, 266]. Microorganisms with substantial resistance to glutaraldehyde have been reported, including some mycobacteria (*M. chelonae*, *Mycobacterium avium-intracellulare, M. xenopi*) [598-601], *Methylobacterium mesophilicum* [602], *Trichosporon*, fungal ascospores (e.g., *Microascus cinereus*, *Cheatomium globosum*), and *Cryptosporidium*[271, 603]. *M. chelonae* persisted in a 0.2% glutaraldehyde solution used to store porcine prosthetic heart valves [604].

Two percent alkaline glutaraldehyde solution inactivated 10^5 *M. tuberculosis* cells on the surface of penicylinders within 5 minutes at 18°C [589]. However, subsequent studies[82] questioned the mycobactericidal prowess of glutaraldehydes. Two percent alkaline glutaraldehyde has slow action (20 to >30 minutes) against *M. tuberculosis* and compares unfavorably with alcohols, formaldehydes, iodine, and phenol [82]. Suspensions of *M. avium, M. intracellulare,* and *M. gordonae* were more resistant to inactivation by a 2% alkaline glutaraldehyde (estimated time to complete inactivation: ~60 minutes) than were virulent *M. tuberculosis* (estimated time to complete inactivation ~25 minutes) [605]. The rate of kill was directly proportional to the temperature, and a standardized suspension of *M. tuberculosis* could not be sterilized within 10 minutes [84]. An FDA-cleared chemical sterilant containing 2.5% glutaraldehyde uses increased temperature (35°C) to reduce the time required to achieve high-level disinfection (5 minutes) [85, 606], but its use is limited to automatic endoscope reprocessors equipped with a heater. In another study employing membrane filters for measurement of mycobactericidal activity of 2% alkaline glutaraldehyde, complete inactivation was achieved within 20 minutes at 20°C when the test inoculum was 10^6 *M. tuberculosis* per membrane [81]. Several investigators [55, 57, 73, 76, 80, 81, 84, 605] have demonstrated that glutaraldehyde solutions inactivate 2.4 to >5.0 $\log_{10}$ of *M. tuberculosis* in 10 minutes (including multidrug-resistant *M. tuberculosis*) and 4.0–6.4 $\log_{10}$ of *M. tuberculosis* in 20 minutes. On the basis of these data and other studies, 20 minutes at room temperature is considered the minimum exposure time needed to reliably kill *Mycobacteria* and other vegetative bacteria with ≥2% glutaraldehyde [17, 19, 27, 57, 83, 94, 108, 111, 117-121, 607].

Glutaraldehyde is commonly diluted during use, and studies showed a glutaraldehyde concentration decline after a few days of use in an automatic endoscope washer [608, 609]. The decline occurs because instruments are not thoroughly dried and water is carried in with the instrument, which increases the solution's volume and dilutes its effective concentration [610]. This emphasizes the need to ensure that semicritical equipment is disinfected with an acceptable concentration of glutaraldehyde. Data suggest that 1.0%–1.5% glutaraldehyde is the minimum effective concentration for >2% glutaraldehyde solutions when used as a high-level disinfectant [76, 589, 590, 609]. Chemical test strips or liquid chemical monitors [610, 611] are available for determining whether an effective concentration of glutaraldehyde is present despite repeated use and dilution. The frequency of testing should be based on how frequently the solutions are used (e.g., used daily, test daily; used weekly, test before use; used 30 times per day, test each 10th use), but the strips should not be used to extend the use life beyond the expiration date. Data suggest the chemicals in the test strip deteriorate with time [612] and a

manufacturer's expiration date should be placed on the bottles. The bottle of test strips should be dated when opened and used for the period of time indicated on the bottle (e.g., 120 days). The results of test strip monitoring should be documented. The glutaraldehyde test kits have been preliminarily evaluated for accuracy and range [612] but the reliability has been questioned [613]. To ensure the presence of minimum effective concentration of the high-level disinfectant, manufacturers of some chemical test strips recommend the use of quality-control procedures to ensure the strips perform properly. If the manufacturer of the chemical test strip recommends a quality-control procedure, users should comply with the manufacturer's recommendations. The concentration should be considered unacceptable or unsafe when the test indicates a dilution below the product's minimum effective concentration (MEC) (generally to ≤1.0%–1.5% glutaraldehyde) by the indicator not changing color.

A 2.0% glutaraldehyde–7.05% phenol–1.20% sodium phenate product that contained 0.125% glutaraldehyde–0.44% phenol–0.075% sodium phenate when diluted 1:16 is not recommended as a high-level disinfectant because it lacks bactericidal activity in the presence of organic matter and lacks tuberculocidal, fungicidal, virucidal, and sporicidal activity [49, 55, 56, 71, 73-79, 614]. In December 1991, EPA issued an order to stop the sale of all batches of this product because of efficacy data showing the product is not effective against spores and possibly other microorganisms or inanimate objects as claimed on the label [615]. FDA has cleared a glutaraldehyde–phenol/phenate concentrate as a high-level disinfectant that contains 1.12% glutaraldehyde with 1.93% phenol/phenate at its use concentration. Other FDA cleared glutaraldehyde sterilants that contain 2.4%–3.4% glutaraldehyde are used undiluted [606].

Uses. Glutaraldehyde is used most commonly as a high-level disinfectant for medical equipment such as endoscopes [69, 107, 504], spirometry tubing, dialyzers [616], transducers, anesthesia and respiratory therapy equipment [617], hemodialysis proportioning and dialysate delivery systems [249, 618], and reuse of laparoscopic disposable plastic trocars [619]. Glutaraldehyde is noncorrosive to metal and does not damage lensed instruments, rubber. or plastics. Glutaraldehyde should not be used for cleaning noncritical surfaces because it is too toxic and expensive.

Colitis believed caused by glutaraldehyde exposure from residual disinfecting solution in endoscope solution channels has been reported and is preventable by careful endoscope rinsing [318, 620-630]. One study found that residual glutaraldehyde levels were higher and more variable after manual disinfection (<0.2 mg/L to 159.5 mg/L) than after automatic disinfection (0.2–6.3 mg/L)[631]. Similarly, keratopathy and corneal decompensation were caused by ophthalmic instruments that were inadequately rinsed after soaking in 2% glutaraldehyde [632, 633].

Healthcare personnel can be exposed to elevated levels of glutaraldehyde vapor when equipment is processed in poorly ventilated rooms, when spills occur, when glutaraldehyde solutions are activated or changed,[634], or when open immersion baths are used. Acute or chronic exposure can result in skin irritation or dermatitis, mucous membrane irritation (eye, nose, mouth), or pulmonary symptoms [318, 635-639]. Epistaxis, allergic contact dermatitis, asthma, and rhinitis also have been reported in healthcare workers exposed to glutaraldehyde [636, 640-647].

Glutaraldehyde exposure should be monitored to ensure a safe work environment. Testing can be done by four techniques: a silica gel tube/gas chromatography with a flame ionization detector, dinitrophenylhydrazine (DNPH)-impregnated filter cassette/high-performance liquid chromatography (HPLC) with an ultraviolet (UV) detector, a passive badge/HPLC, or a handheld glutaraldehyde air monitor [648]. The silica gel tube and the DNPH-impregnated cassette are suitable for monitoring the 0.05 ppm ceiling limit. The passive badge, with a 0.02 ppm limit of detection, is considered marginal at the Americal Council of Governmental Industrial Hygienists (ACGIH) ceiling level. The ceiling level is considered too close to the glutaraldehyde meter's 0.03 ppm limit of detection to provide confidence in the readings [648]. ACGIH does not require a specific monitoring schedule for glutaraldehyde; however, a monitoring schedule is needed to ensure the level is less than the ceiling limit. For example, monitoring

should be done initially to determine glutaraldehyde levels, after procedural or equipment changes, and in response to worker complaints [649]. In the absence of an OSHA permissible exposure limit, if the glutaraldehyde level is higher than the ACGIH ceiling limit of 0.05 ppm, corrective action and repeat monitoring would be prudent [649].

Engineering and work-practice controls that can be used to resolve these problems include ducted exhaust hoods, air systems that provide 7–15 air exchanges per hour, ductless fume hoods with absorbents for the glutaraldehyde vapor, tight-fitting lids on immersion baths, personal protection (e.g., nitrile or butyl rubber gloves but not natural latex gloves, goggles) to minimize skin or mucous membrane contact, and automated endoscope processors [7, 650]. If engineering controls fail to maintain levels below the ceiling limit, institutions can consider the use of respirators (e.g., a half-face respirator with organic vapor cartridge [640] or a type "C" supplied air respirator with a full facepiece operated in a positive pressure mode) [651]. In general, engineering controls are preferred over work-practice and administrative controls because they do not require active participation by the health-care worker. Even though enforcement of the OSHA ceiling limit was suspended in 1993 by the U.S. Court of Appeals [577], limiting employee exposure to 0.05 ppm (according to ACGIH) is prudent because, at this level, glutaraldehyde can irritate the eyes, throat, and nose [318, 577, 639, 652]. If glutaraldehyde disposal through the sanitary sewer system is restricted, sodium bisulfate can be used to neutralize the glutaraldehyde and make it safe for disposal.

Hydrogen Peroxide

Overview. The literature contains several accounts of the properties, germicidal effectiveness, and potential uses for stabilized hydrogen peroxide in the health-care setting. Published reports ascribe good germicidal activity to hydrogen peroxide and attest to its bactericidal, virucidal, sporicidal, and fungicidal properties [653-655]. (Tables 4 and 5) The FDA website lists cleared liquid chemical sterilants and high-level disinfectants containing hydrogen peroxide and their cleared contact conditions.

Mode of Action. Hydrogen peroxide works by producing destructive hydroxyl free radicals that can attack membrane lipids, DNA, and other essential cell components. Catalase, produced by aerobic organisms and facultative anaerobes that possess cytochrome systems, can protect cells from metabolically produced hydrogen peroxide by degrading hydrogen peroxide to water and oxygen. This defense is overwhelmed by the concentrations used for disinfection [653, 654].

Microbicidal Activity. Hydrogen peroxide is active against a wide range of microorganisms, including bacteria, yeasts, fungi, viruses, and spores [78, 654]. A 0.5% accelerated hydrogen peroxide demonstrated bactericidal and virucidal activity in 1 minute and mycobactericidal and fungicidal activity in 5 minutes [656]. Bactericidal effectiveness and stability of hydrogen peroxide in urine has been demonstrated against a variety of health-care–associated pathogens; organisms with high cellular catalase activity (e.g., *S. aureus*, *S. marcescens*, and *Proteus mirabilis*) required 30–60 minutes of exposure to 0.6% hydrogen peroxide for a 10^8 reduction in cell counts, whereas organisms with lower catalase activity (e.g., *E. coli*, *Streptococcus* species, and *Pseudomonas* species) required only 15 minutes' exposure [657]. In an investigation of 3%, 10%, and 15% hydrogen peroxide for reducing spacecraft bacterial populations, a complete kill of 10^6 spores (i.e., *Bacillus* species) occurred with a 10% concentration and a 60-minute exposure time. A 3% concentration for 150 minutes killed 10^6 spores in six of seven exposure trials [658]. A 10% hydrogen peroxide solution resulted in a 10^3 decrease in *B. atrophaeus* spores, and a $\geq 10^5$ decrease when tested against 13 other pathogens in 30 minutes at 20°C [659, 660]. A 3.0% hydrogen peroxide solution was ineffective against VRE after 3 and 10 minutes exposure times [661] and caused only a 2-$\log_{10}$ reduction in the number of *Acanthamoeba* cysts in approximately 2 hours [662]. A 7% stabilized hydrogen peroxide proved to be sporicidal (6 hours of exposure), mycobactericidal (20 minutes), fungicidal (5 minutes) at full strength, virucidal (5 minutes) and bactericidal (3 minutes) at a 1:16 dilution when a quantitative carrier test was used [655]. The 7% solution of hydrogen peroxide, tested after 14 days of stress (in the form of germ-loaded carriers and respiratory therapy equipment), was sporicidal (>7 $\log_{10}$ reduction in 6 hours), mycobactericidal (>6.5 $\log_{10}$ reduction in 25

minutes), fungicidal (>5 $\log_{10}$ reduction in 20 minutes), bactericidal (>6 $\log_{10}$ reduction in 5 minutes) and virucidal (5 $\log_{10}$ reduction in 5 minutes) [663]. Synergistic sporicidal effects were observed when spores were exposed to a combination of hydrogen peroxide (5.9%–23.6%) and peracetic acid [664]. Other studies demonstrated the antiviral activity of hydrogen peroxide against rhinovirus [665]. The time required for inactivating three serotypes of rhinovirus using a 3% hydrogen peroxide solution was 6–8 minutes; this time increased with decreasing concentrations (18-20 minutes at 1.5%, 50–60 minutes at 0.75%).

Concentrations of hydrogen peroxide from 6% to 25% show promise as chemical sterilants. The product marketed as a sterilant is a premixed, ready-to-use chemical that contains 7.5% hydrogen peroxide and 0.85% phosphoric acid (to maintain a low pH) [69]. The mycobactericidal activity of 7.5% hydrogen peroxide has been corroborated in a study showing the inactivation of >10^5 multidrug-resistant *M. tuberculosis* after a 10-minute exposure [666]. Thirty minutes were required for >99.9% inactivation of poliovirus and HAV [667]. Three percent and 6% hydrogen peroxide were unable to inactivate HAV in 1 minute in a carrier test [58]. When the effectiveness of 7.5% hydrogen peroxide at 10 minutes was compared with 2% alkaline glutaraldehyde at 20 minutes in manual disinfection of endoscopes, no significant difference in germicidal activity was observed [668].). No complaints were received from the nursing or medical staff regarding odor or toxicity. In one study, 6% hydrogen peroxide (unused product was 7.5%) was more effective in the high-level disinfection of flexible endoscopes than was the 2% glutaraldehyde solution [456]. A new, rapid-acting 13.4% hydrogen peroxide formulation (that is not yet FDA-cleared) has demonstrated sporicidal, mycobactericidal, fungicidal, and virucidal efficacy. Manufacturer data demonstrate that this solution sterilizes in 30 minutes and provides high-level disinfection in 5 minutes[669]. This product has not been used long enough to evaluate material compatibility to endoscopes and other semicritical devices, and further assessment by instrument manufacturers is needed.

Under normal conditions, hydrogen peroxide is extremely stable when properly stored (e.g., in dark containers). The decomposition or loss of potency in small containers is less than 2% per year at ambient temperatures [670].

Uses. Commercially available 3% hydrogen peroxide is a stable and effective disinfectant when used on inanimate surfaces. It has been used in concentrations from 3% to 6% for disinfecting soft contact lenses (e.g., 3% for 2–3 hrs) [653, 671, 672], tonometer biprisms [513], ventilators [673], fabrics [397], and endoscopes [456]. Hydrogen peroxide was effective in spot-disinfecting fabrics in patients' rooms [397]. Corneal damage from a hydrogen peroxide-soaked tonometer tip that was not properly rinsed has been reported [674]. Hydrogen peroxide also has been instilled into urinary drainage bags in an attempt to eliminate the bag as a source of bladder bacteriuria and environmental contamination [675]. Although the instillation of hydrogen peroxide into the bag reduced microbial contamination of the bag, this procedure did not reduce the incidence of catheter-associated bacteriuria [675].

A chemical irritation resembling pseudomembranous colitis caused by either 3% hydrogen peroxide or a 2% glutaraldehyde has been reported [621]. An epidemic of pseudomembrane-like enteritis and colitis in seven patients in a gastrointestinal endoscopy unit also has been associated with inadequate rinsing of 3% hydrogen peroxide from the endoscope [676].

As with other chemical sterilants, dilution of the hydrogen peroxide must be monitored by regularly testing the minimum effective concentration (i.e., 7.5%–6.0%). Compatibility testing by Olympus America of the 7.5% hydrogen peroxide found both cosmetic changes (e.g., discoloration of black anodized metal finishes) [69] and functional changes with the tested endoscopes (Olympus, written communication, October 15, 1999).

Iodophors

Overview. Iodine solutions or tinctures long have been used by health professionals primarily as antiseptics on skin or tissue. Iodophors, on the other hand, have been used both as antiseptics and

disinfectants. FDA has not cleared any liquid chemical sterilant or high-level disinfectants with iodophors as the main active ingredient. An iodophor is a combination of iodine and a solubilizing agent or carrier; the resulting complex provides a sustained-release reservoir of iodine and releases small amounts of free iodine in aqueous solution. The best-known and most widely used iodophor is povidone-iodine, a compound of polyvinylpyrrolidone with iodine. This product and other iodophors retain the germicidal efficacy of iodine but unlike iodine generally are nonstaining and relatively free of toxicity and irritancy [677, 678].

Several reports that documented intrinsic microbial contamination of antiseptic formulations of povidone-iodine and poloxamer-iodine [679-681] caused a reappraisal of the chemistry and use of iodophors[682]. "Free" iodine (I_2) contributes to the bactericidal activity of iodophors and dilutions of iodophors demonstrate more rapid bactericidal action than does a full-strength povidone-iodine solution. The reason for the observation that dilution increases bactericidal activity is unclear, but dilution of povidone-iodine might weaken the iodine linkage to the carrier polymer with an accompanying increase of free iodine in solution [680]. Therefore, iodophors must be diluted according to the manufacturers' directions to achieve antimicrobial activity.

Mode of Action. Iodine can penetrate the cell wall of microorganisms quickly, and the lethal effects are believed to result from disruption of protein and nucleic acid structure and synthesis.

Microbicidal Activity. Published reports on the in vitro antimicrobial efficacy of iodophors demonstrate that iodophors are bactericidal, mycobactericidal, and virucidal but can require prolonged contact times to kill certain fungi and bacterial spores [14, 71-73, 290, 683-686]. Three brands of povidone-iodine solution have demonstrated more rapid kill (seconds to minutes) of *S. aureus* and *M. chelonae* at a 1:100 dilution than did the stock solution [683]. The virucidal activity of 75–150 ppm available iodine was demonstrated against seven viruses [72]. Other investigators have questioned the efficacy of iodophors against poliovirus in the presence of organic matter [685]and rotavirus SA-11 in distilled or tapwater [290]. Manufacturers' data demonstrate that commercial iodophors are not sporicidal, but they are tuberculocidal, fungicidal, virucidal, and bactericidal at their recommended use-dilution.

Uses. Besides their use as an antiseptic, iodophors have been used for disinfecting blood culture bottles and medical equipment, such as hydrotherapy tanks, thermometers, and endoscopes. Antiseptic iodophors are not suitable for use as hard-surface disinfectants because of concentration differences. Iodophors formulated as antiseptics contain less free iodine than do those formulated as disinfectants [376]. Iodine or iodine-based antiseptics should not be used on silicone catheters because they can adversely affect the silicone tubing [687].

Ortho-phthalaldehyde (OPA)

Overview. Ortho-phthalaldehyde is a high-level disinfectant that received FDA clearance in October 1999. It contains 0.55% 1,2-benzenedicarboxaldehyde (OPA). OPA solution is a clear, pale-blue liquid with a pH of 7.5. (Tables 4 and 5)

***Mode of Action*.** Preliminary studies on the mode of action of OPA suggest that both OPA and glutaraldehyde interact with amino acids, proteins, and microorganisms. However, OPA is a less potent cross-linking agent. This is compensated for by the lipophilic aromatic nature of OPA that is likely to assist its uptake through the outer layers of mycobacteria and gram-negative bacteria [688-690]. OPA appears to kill spores by blocking the spore germination process [691].

***Microbicidal Activity*.** Studies have demonstrated excellent microbicidal activity in vitro [69, 100, 271, 400, 692-703]. For example, OPA has superior mycobactericidal activity (5-$\log_{10}$ reduction in 5 minutes) to glutaraldehyde. The mean times required to produce a 6-$\log_{10}$ reduction for *M. bovis* using 0.21% OPA was 6 minutes, compared with 32 minutes using 1.5% glutaraldehyde [693]. OPA showed good activity against the mycobacteria tested, including the glutaraldehyde-resistant strains, but 0.5% OPA was not sporicidal with 270 minutes of exposure. Increasing the pH from its unadjusted level (about 6.5) to pH 8 improved the sporicidal activity of OPA [694]. The level of biocidal activity was directly related to the

temperature. A greater than 5-$\log_{10}$ reduction of *B. atrophaeus* spores was observed in 3 hours at 35°C, than in 24 hours at 20°C. Also, with an exposure time ≤5 minutes, biocidal activity decreased with increasing serum concentration. However, efficacy did not differ when the exposure time was ≥10 minutes [697]. In addition, OPA is effective (>5-$\log_{10}$ reduction) against a wide range of microorganisms, including glutaraldehyde-resistant mycobacteria and *B. atrophaeus* spores [694].

The influence of laboratory adaptation of test strains, such as *P. aeruginosa*, to 0.55% OPA has been evaluated. Resistant and multiresistant strains increased substantially in susceptibility to OPA after laboratory adaptation ($\log_{10}$ reduction factors increased by 0.54 and 0.91 for resistant and multiresistant strains, respectively) [704]. Other studies have found naturally occurring cells of *P. aeurginosa* were more resistant to a variety of disinfectants than were subcultured cells [705].

Uses. OPA has several potential advantages over glutaraldehyde. It has excellent stability over a wide pH range (pH 3–9), is not a known irritant to the eyes and nasal passages [706], does not require exposure monitoring, has a barely perceptible odor, and requires no activation. OPA, like glutaraldehyde, has excellent material compatibility. A potential disadvantage of OPA is that it stains proteins gray (including unprotected skin) and thus must be handled with caution [69]. However, skin staining would indicate improper handling that requires additional training and/or personal protective equipment (e.g., gloves, eye and mouth protection, and fluid-resistant gowns). OPA residues remaining on inadequately water-rinsed transesophageal echo probes can stain the patient's mouth [707]. Meticulous cleaning, using the correct OPA exposure time (e.g., 12 minutes) and copious rinsing of the probe with water should eliminate this problem. The results of one study provided a basis for a recommendation that rinsing of instruments disinfected with OPA will require at least 250 mL of water per channel to reduce the chemical residue to a level that will not compromise patient or staff safety (<1 ppm) [708]. Personal protective equipment should be worn when contaminated instruments, equipment, and chemicals are handled [400]. In addition, equipment must be thoroughly rinsed to prevent discoloration of a patient's skin or mucous membrane.

In April 2004, the manufacturer of OPA disseminated information to users about patients who reportedly experienced an anaphylaxis-like reaction after cystoscopy where the scope had been reprocessed using OPA. Of approximately 1 million urologic procedures performed using instruments reprocessed using OPA, 24 cases (17 cases in the United States, six in Japan, one in the United Kingdom) of anaphylaxis-like reactions have been reported after repeated cystoscopy (typically after four to nine treatments). Preventive measures include removal of OPA residues by thorough rinsing and not using OPA for reprocessing urologic instrumentation used to treat patients with a history of bladder cancer (Nevine Erian, personal communication, June 4, 2004; Product Notification, Advanced Sterilization Products, April 23, 2004) [709].

A few OPA clinical studies are available. In a clinical-use study, OPA exposure of 100 endoscopes for 5 minutes resulted in a >5-$\log_{10}$ reduction in bacterial load. Furthermore, OPA was effective over a 14-day use cycle [100]. Manufacturer data show that OPA will last longer in an automatic endoscope reprocessor before reaching its MEC limit (MEC after 82 cycles) than will glutaraldehyde (MEC after 40 cycles) [400]. High-pressure liquid chromatography confirmed that OPA levels are maintained above 0.3% for at least 50 cycles [706, 710]. OPA must be disposed in accordance with local and state regulations. If OPA disposal through the sanitary sewer system is restricted, glycine (25 grams/gallon) can be used to neutralize the OPA and make it safe for disposal.

The high-level disinfectant label claims for OPA solution at 20°C vary worldwide (e.g., 5 minutes in Europe, Asia, and Latin America; 10 minutes in Canada and Australia; and 12 minutes in the United States). These label claims differ worldwide because of differences in the test methodology and requirements for licensure. In an automated endoscope reprocessor with an FDA-cleared capability to maintain solution temperatures at 25°C, the contact time for OPA is 5 minutes.

Peracetic Acid

Overview. Peracetic, or peroxyacetic, acid is characterized by rapid action against all microorganisms. Special advantages of peracetic acid are that it lacks harmful decomposition products (i.e., acetic acid, water, oxygen, hydrogen peroxide), enhances removal of organic material [711], and leaves no residue. It remains effective in the presence of organic matter and is sporicidal even at low temperatures (Tables 4 and 5). Peracetic acid can corrode copper, brass, bronze, plain steel, and galvanized iron but these effects can be reduced by additives and pH modifications. It is considered unstable, particularly when diluted; for example, a 1% solution loses half its strength through hydrolysis in 6 days, whereas 40% peracetic acid loses 1%–2% of its active ingredients per month [654].

***Mode of Action*.** Little is known about the mechanism of action of peracetic acid, but it is believed to function similarly to other oxidizing agents—that is, it denatures proteins, disrupts the cell wall permeability, and oxidizes sulfhydryl and sulfur bonds in proteins, enzymes, and other metabolites [654].

***Microbicidal Activity*.** Peracetic acid will inactivate gram-positive and gram-negative bacteria, fungi, and yeasts in ≤5 minutes at <100 ppm. In the presence of organic matter, 200–500 ppm is required. For viruses, the dosage range is wide (12–2250 ppm), with poliovirus inactivated in yeast extract in 15 minutes with 1,500–2,250 ppm. In one study, 3.5% peracetic acid was ineffective against HAV after 1-minute exposure using a carrier test [58]. Peracetic acid (0.26%) was effective ($\log_{10}$ reduction factor >5) against all test strains of mycobacteria (*M. tuberculosis, M. avium-intracellulare, M. chelonae, and M. fortuitum*) within 20–30 minutes in the presence or absence of an organic load [607, 712]. With bacterial spores, 500–10,000 ppm (0.05%–1%) inactivates spores in 15 seconds to 30 minutes using a spore suspension test [654, 659, 713-715].

Uses. An automated machine using peracetic acid to chemically sterilize medical (e.g., endoscopes, arthroscopes), surgical, and dental instruments is used in the United States[716-718]. As previously noted, dental handpieces should be steam sterilized. The sterilant, 35% peracetic acid, is diluted to 0.2% with filtered water at 50°C. Simulated-use trials have demonstrated excellent microbicidal activity [111, 718-722], and three clinical trials have demonstrated both excellent microbial killing and no clinical failures leading to infection[90, 723, 724]. The high efficacy of the system was demonstrated in a comparison of the efficacies of the system with that of ethylene oxide. Only the peracetic acid system completely killed 6 $\log_{10}$ of *M. chelonae, E. faecalis,* and *B. atrophaeus* spores with both an organic and inorganic challenge[722]. An investigation that compared the costs, performance, and maintenance of urologic endoscopic equipment processed by high-level disinfection (with glutaraldehyde) with those of the peracetic acid system reported no clinical differences between the two systems. However, the use of this system led to higher costs than the high-level disinfection, including costs for processing ($6.11 vs. $0.45 per cycle), purchasing and training ($24,845 vs. $16), installation ($5,800 vs. $0), and endoscope repairs ($6,037 vs. $445) [90]. Furthermore, three clusters of infection using the peracetic acid automated endoscope reprocessor were linked to inadequately processed bronchoscopes when inappropriate channel connectors were used with the system [725]. These clusters highlight the importance of training, proper model-specific endoscope connector systems, and quality-control procedures to ensure compliance with endoscope manufacturer recommendations and professional organization guidelines. An alternative high-level disinfectant available in the United Kingdom contains 0.35% peracetic acid. Although this product is rapidly effective against a broad range of microorganisms [466, 726, 727], it tarnishes the metal of endoscopes and is unstable, resulting in only a 24-hour use life [727].

Peracetic Acid and Hydrogen Peroxide

Overview. Two chemical sterilants are available that contain peracetic acid plus hydrogen peroxide (i.e., 0.08% peracetic acid plus 1.0% hydrogen peroxide [no longer marketed]; and 0.23% peracetic acid plus 7.35% hydrogen peroxide (Tables 4 and 5).

Microbicidal Activity. The bactericidal properties of peracetic acid and hydrogen peroxide have been demonstrated [728]. Manufacturer data demonstrated this combination of peracetic acid and

hydrogen peroxide inactivated all microorganisms except bacterial spores within 20 minutes. The 0.08% peracetic acid plus 1.0% hydrogen peroxide product effectively inactivated glutaraldehyde-resistant mycobacteria[729].

Uses. The combination of peracetic acid and hydrogen peroxide has been used for disinfecting hemodialyzers [730]. The percentage of dialysis centers using a peracetic acid-hydrogen peroxide-based disinfectant for reprocessing dialyzers increased from 5% in 1983 to 56% in 1997[249]. Olympus America does not endorse use of 0.08% peracetic acid plus 1.0% hydrogen peroxide (Olympus America, personal communication, April 15, 1998) on any Olympus endoscope because of cosmetic and functional damage and will not assume liability for chemical damage resulting from use of this product. This product is not currently available. FDA has cleared a newer chemical sterilant with 0.23% peracetic acid and 7.35% hydrogen peroxide (Tables 4 and 5). After testing the 7.35% hydrogen peroxide and 0.23% peracetic acid product, Olympus America concluded it was not compatible with the company's flexible gastrointestinal endoscopes; this conclusion was based on immersion studies where the test insertion tubes had failed because of swelling and loosening of the black polymer layer of the tube (Olympus America, personal communication, September 13, 2000).

Phenolics

Overview. Phenol has occupied a prominent place in the field of hospital disinfection since its initial use as a germicide by Lister in his pioneering work on antiseptic surgery. In the past 30 years, however, work has concentrated on the numerous phenol derivatives or phenolics and their antimicrobial properties. Phenol derivatives originate when a functional group (e.g., alkyl, phenyl, benzyl, halogen) replaces one of the hydrogen atoms on the aromatic ring. Two phenol derivatives commonly found as constituents of hospital disinfectants are *ortho*-phenylphenol and *ortho*-benzyl-*para*-chlorophenol. The antimicrobial properties of these compounds and many other phenol derivatives are much improved over those of the parent chemical. Phenolics are absorbed by porous materials, and the residual disinfectant can irritate tissue. In 1970, depigmentation of the skin was reported to be caused by phenolic germicidal detergents containing *para*-tertiary butylphenol and *para*-tertiary amylphenol [731].

Mode of Action. In high concentrations, phenol acts as a gross protoplasmic poison, penetrating and disrupting the cell wall and precipitating the cell proteins. Low concentrations of phenol and higher molecular-weight phenol derivatives cause bacterial death by inactivation of essential enzyme systems and leakage of essential metabolites from the cell wall [732].

Microbicidal Activity. Published reports on the antimicrobial efficacy of commonly used phenolics showed they were bactericidal, fungicidal, virucidal, and tuberculocidal [14, 61, 71, 73, 227, 416, 573, 732-738]. One study demonstrated little or no virucidal effect of a phenolic against coxsackie B4, echovirus 11, and poliovirus 1 [736]. Similarly, 12% *ortho*-phenylphenol failed to inactivate any of the three hydrophilic viruses after a 10-minute exposure time, although 5% phenol was lethal for these viruses [72]. A 0.5% dilution of a phenolic (2.8% *ortho*-phenylphenol and 2.7% *ortho*-benzyl-*para*-chlorophenol) inactivated HIV [227] and a 2% solution of a phenolic (15% ortho-phenylphenol and 6.3% para-tertiary-amylphenol) inactivated all but one of 11 fungi tested [71].

Manufacturers' data using the standardized AOAC methods demonstrate that commercial phenolics are not sporicidal but are tuberculocidal, fungicidal, virucidal, and bactericidal at their recommended use-dilution. Attempts to substantiate the bactericidal label claims of phenolics using the AOAC Use-Dilution Method occasionally have failed [416, 737]. However, results from these same studies have varied dramatically among laboratories testing identical products.

Uses. Many phenolic germicides are EPA-registered as disinfectants for use on environmental surfaces (e.g., bedside tables, bedrails, and laboratory surfaces) and noncritical medical devices. Phenolics are not FDA-cleared as high-level disinfectants for use with semicritical items but could be used to preclean or decontaminate critical and semicritical devices before terminal sterilization or high-

level disinfection.

The use of phenolics in nurseries has been questioned because of hyperbilirubinemia in infants placed in bassinets where phenolic detergents were used [739]. In addition, bilirubin levels were reported to increase in phenolic-exposed infants, compared with nonphenolic-exposed infants, when the phenolic was prepared according to the manufacturers' recommended dilution [740]. If phenolics are used to clean nursery floors, they must be diluted as recommended on the product label. Phenolics (and other disinfectants) should not be used to clean infant bassinets and incubators while occupied. If phenolics are used to terminally clean infant bassinets and incubators, the surfaces should be rinsed thoroughly with water and dried before reuse of infant bassinets and incubators [17].

Quaternary Ammonium Compounds

Overview. The quaternary ammonium compounds are widely used as disinfectants. Health-care–associated infections have been reported from contaminated quaternary ammonium compounds used to disinfect patient-care supplies or equipment, such as cystoscopes or cardiac catheters [741, 742]. The quaternaries are good cleaning agents, but high water hardness [743] and materials such as cotton and gauze pads can make them less microbicidal because of insoluble precipitates or cotton and gauze pads absorb the active ingredients, respectively. One study showed a significant decline (~40%–50% lower at 1 hour) in the concentration of quaternaries released when cotton rags or cellulose-based wipers were used in the open-bucket system, compared with the nonwoven spunlace wipers in the closed-bucket system [744] As with several other disinfectants (e.g., phenolics, iodophors) gram-negative bacteria can survive or grow in them [404].

Chemically, the quaternaries are organically substituted ammonium compounds in which the nitrogen atom has a valence of 5, four of the substituent radicals (R1-R4) are alkyl or heterocyclic radicals of a given size or chain length, and the fifth (X^-) is a halide, sulfate, or similar radical [745]. Each compound exhibits its own antimicrobial characteristics, hence the search for one compound with outstanding antimicrobial properties. Some of the chemical names of quaternary ammonium compounds used in healthcare are alkyl dimethyl benzyl ammonium chloride, alkyl didecyl dimethyl ammonium chloride, and dialkyl dimethyl ammonium chloride. The newer quaternary ammonium compounds (i.e., fourth generation), referred to as twin-chain or dialkyl quaternaries (e.g. didecyl dimethyl ammonium bromide and dioctyl dimethyl ammonium bromide), purportedly remain active in hard water and are tolerant of anionic residues [746].

A few case reports have documented occupational asthma as a result of exposure to benzalkonium chloride [747].

Mode of Action. The bactericidal action of the quaternaries has been attributed to the inactivation of energy-producing enzymes, denaturation of essential cell proteins, and disruption of the cell membrane[746]. Evidence exists that supports these and other possibilities [745 748].

Microbicidal Activity. Results from manufacturers' data sheets and from published scientific literature indicate that the quaternaries sold as hospital disinfectants are generally fungicidal, bactericidal, and virucidal against lipophilic (enveloped) viruses; they are not sporicidal and generally not tuberculocidal or virucidal against hydrophilic (nonenveloped) viruses[14, 54-56, 58, 59, 61, 71, 73, 186, 297, 748, 749]. The poor mycobactericidal activities of quaternary ammonium compounds have been demonstrated [55, 73]. Quaternary ammonium compounds (as well as 70% isopropyl alcohol, phenolic, and a chlorine-containing wipe [80 ppm]) effectively (>95%) remove and/or inactivate contaminants (i.e., multidrug-resistant *S. aureus*, vancomycin-resistant *Entercoccus*, *P. aeruginosa*) from computer keyboards with a 5-second application time. No functional damage or cosmetic changes occurred to the computer keyboards after 300 applications of the disinfectants [45].

Attempts to reproduce the manufacturers' bactericidal and tuberculocidal claims using the AOAC

tests with a limited number of quaternary ammonium compounds occasionally have failed [73, 416, 737]. However, test results have varied extensively among laboratories testing identical products [416, 737].

Uses. The quaternaries commonly are used in ordinary environmental sanitation of noncritical surfaces, such as floors, furniture, and walls. EPA-registered quaternary ammonium compounds are appropriate to use for disinfecting medical equipment that contacts intact skin (e.g., blood pressure cuffs).

MISCELLANEOUS INACTIVATING AGENTS

Other Germicides

Several compounds have antimicrobial activity but for various reasons have not been incorporated into the armamentarium of health-care disinfectants. These include mercurials, sodium hydroxide, β-propiolactone, chlorhexidine gluconate, cetrimide-chlorhexidine, glycols (triethylene and propylene), and the Tego disinfectants. Two authoritative references examine these agents in detail [16, 412].

A peroxygen-containing formulation had marked bactericidal action when used as a 1% weight/volume solution and virucidal activity at 3% [49], but did not have mycobactericidal activity at concentrations of 2.3% and 4% and exposure times ranging from 30 to 120 minutes [750]. It also required 20 hours to kill *B. atrophaeus* spores [751]. A powder-based peroxygen compound for disinfecting contaminated spill was strongly and rapidly bactericidal [752].

In preliminary studies, nanoemulsions (composed of detergents and lipids in water) showed activity against vegetative bacteria, enveloped viruses and *Candida*. This product represents a potential agent for use as a topical biocidal agent. [753-755].

New disinfectants that require further evaluation include glucoprotamin[756], tertiary amines [703]. and a light-activated antimicrobial coating [757]. Several other disinfection technologies might have potential applications in the healthcare setting [758].

Metals as Microbicides

Comprehensive reviews of antisepsis [759], disinfection[421], and anti-infective chemotherapy [760] barely mention the antimicrobial activity of heavy metals[761, 762]. Nevertheless, the anti-infective activity of some heavy metals has been known since antiquity. Heavy metals such as silver have been used for prophylaxis of conjunctivitis of the newborn, topical therapy for burn wounds, and bonding to indwelling catheters, and the use of heavy metals as antiseptics or disinfectants is again being explored [763]. Inactivation of bacteria on stainless steel surfaces by zeolite ceramic coatings containing silver and zinc ions has also been demonstrated [764, 765].

Metals such as silver, iron, and copper could be used for environmental control, disinfection of water, or reusable medical devices or incorporated into medical devices (e.g., intravascular catheters) [400, 761-763, 766-770]. A comparative evaluation of six disinfectant formulations for residual antimicrobial activity demonstrated that only the silver disinfectant demonstrated significant residual activity against *S. aureus* and *P. aeruginosa* [763]. Preliminary data suggest metals are effective against a wide variety of microorganisms.

Clinical uses of other heavy metals include copper-8-quinolinolate as a fungicide against *Aspergillus*, copper-silver ionization for *Legionella* disinfection [771-774], organic mercurials as an antiseptic (e.g., mercurochrome) and preservative/disinfectant (e.g., thimerosal [currently being removed from vaccines]) in pharmaceuticals and cosmetics [762].

Ultraviolet Radiation (UV)

The wavelength of UV radiation ranges from 328 nm to 210 nm (3280 A to 2100 A). Its maximum bactericidal effect occurs at 240–280 nm. Mercury vapor lamps emit more than 90% of their radiation at 253.7 nm, which is near the maximum microbicidal activity [775]. Inactivation of microorganisms results from destruction of nucleic acid through induction of thymine dimers. UV radiation has been employed in the disinfection of drinking water [776], air [775], titanium implants [777], and contact lenses[778]. Bacteria and viruses are more easily killed by UV light than are bacterial spores [775]. UV radiation has several potential applications, but unfortunately its germicidal effectiveness and use is influenced by organic matter; wavelength; type of suspension; temperature; type of microorganism; and UV intensity, which is affected by distance and dirty tubes[779]. The application of UV radiation in the health-care environment (i.e.,

operating rooms, isolation rooms, and biologic safety cabinets) is limited to destruction of airborne organisms or inactivation of microorganisms on surfaces. The effect of UV radiation on postoperative wound infections was investigated in a double-blind, randomized study in five university medical centers. After following 14,854 patients over a 2-year period, the investigators reported the overall wound infection rate was unaffected by UV radiation, although postoperative infection in the "refined clean" surgical procedures decreased significantly (3.8%–2.9%) [780]. No data support the use of UV lamps in isolation rooms, and this practice has caused at least one epidemic of UV-induced skin erythema and keratoconjunctivitis in hospital patients and visitors [781].

Pasteurization

Pasteurization is not a sterilization process; its purpose is to destroy all pathogenic microorganisms. However, pasteurization does not destroy bacterial spores. The time-temperature relation for hot-water pasteurization is generally ~70°C (158°F) for 30 minutes. The water temperature and time should be monitored as part of a quality-assurance program [782]. Pasteurization of respiratory therapy [783, 784] and anesthesia equipment [785]is a recognized alternative to chemical disinfection. The efficacy of this process has been tested using an inoculum that the authors believed might simulate contamination by an infected patient. Use of a large inoculum (10^7) of *P. aeruginosa* or *Acinetobacter calcoaceticus* in sets of respiratory tubing before processing demonstrated that machine-assisted chemical processing was more efficient than machine-assisted pasteurization with a disinfection failure rate of 6% and 83%, respectively [783]. Other investigators found hot water disinfection to be effective (inactivation factor >5 $\log_{10}$) against multiple bacteria, including multidrug-resistant bacteria, for disinfecting reusable anesthesia or respiratory therapy equipment [784-786].

Flushing- and Washer-Disinfectors

Flushing- and washer-disinfectors are automated and closed equipment that clean and disinfect objects from bedpans and washbowls to surgical instruments and anesthesia tubes. Items such as bedpans and urinals can be cleaned and disinfected in flushing-disinfectors. They have a short cycle of a few minutes. They clean by flushing with warm water, possibly with a detergent, and then disinfect by flushing the items with hot water or with steam. Because this machine empties, cleans, and disinfects, manual cleaning is eliminated, fewer disposable items are needed, and fewer chemical germicides are used. A microbiologic evaluation of one washer/disinfector demonstrated complete inactivation of suspensions of *E. faecalis* or poliovirus [787]. Other studies have shown that strains of *Enterococcus faecium* can survive the British Standard for heat disinfection of bedpans (80°C for 1 minute). The significance of this finding with reference to the potential for enterococci to survive and disseminate in the health-care environment is debatable [788-790]. These machines are available and used in many European countries.

Surgical instruments and anesthesia equipment are more difficult to clean. They are run in washer-disinfectors on a longer cycle of approximately 20–30 minutes with a detergent. These machines also disinfect by hot water at approximately 90°C [791].

THE REGULATORY FRAMEWORK FOR DISINFECTANTS AND STERILANTS

Before using the guidance provided in this document, health-care workers should be aware of the federal laws and regulations that govern the sale, distribution, and use of disinfectants and sterilants. In particular, health-care workers need to know what requirements pertain to them when they apply these products. Finally, they should understand the relative roles of EPA, FDA, and CDC so the context for the guidance provided in this document is clear.

EPA and FDA

In the United States, chemical germicides formulated as sanitizers, disinfectants, or sterilants are regulated in interstate commerce by the Antimicrobials Division, Office of Pesticides Program, EPA, under the authority of the Federal Insecticide, Fungicide, and Rodenticide Act (FIFRA) of 1947, as amended [792]. Under FIFRA, any substance or mixture of substances intended to prevent, destroy, repel, or mitigate any pest (including microorganisms but excluding those in or on living humans or animals) must be registered before sale or distribution. To obtain a registration, a manufacturer must submit specific data about the safety and effectiveness of each product. For example, EPA requires manufacturers of sanitizers, disinfectants, or chemical sterilants to test formulations by using accepted methods for microbiocidal activity, stability, and toxicity to animals and humans. The manufacturers submit these data to EPA along with proposed labeling. If EPA concludes the product can be used without causing "unreasonable adverse effects," then the product and its labeling are registered, and the manufacturer can sell and distribute the product in the United States.

FIFRA also requires users of products to follow explicitly the labeling directions on each product. The following standard statement appears on all labels under the "Directions for Use" heading: "It is a violation of federal law to use this product in a manner inconsistent with its labeling." This statement means a health-care worker must follow the safety precautions and use directions on the labeling of each registered product. Failure to follow the specified use-dilution, contact time, method of application, or any other condition of use is considered a misuse of the product and potentially subject to enforcement action under FIFRA.

In general, EPA regulates disinfectants and sterilants used on environmental surfaces, and not those used on critical or semicritical medical devices; the latter are regulated by FDA. In June 1993, FDA and EPA issued a "Memorandum of Understanding" that divided responsibility for review and surveillance of chemical germicides between the two agencies. Under the agreement, FDA regulates liquid chemical sterilants used on critical and semicritical devices, and EPA regulates disinfectants used on noncritical surfaces and gaseous sterilants [793]. In 1996, Congress passed the Food Quality Protection Act (FQPA). This act amended FIFRA in regard to several types of products regulated by both EPA and FDA. One provision of FQPA removed regulation of liquid chemical sterilants used on critical and semicritical medical devices from EPA's jurisdiction, and it now rests solely with FDA [792, 794]. EPA continues to register nonmedical chemical sterilants. FDA and EPA have considered the impact of FQPA, and in January 2000, FDA published its final guidance document on product submissions and labeling. Antiseptics are considered antimicrobial drugs used on living tissue and thus are regulated by FDA under the Food, Drug and Cosmetic Act. FDA regulates liquid chemical sterilants and high-level disinfectants intended to process critical and semicritical devices. FDA has published recommendations on the types of test methods that manufacturers should submit to FDA for 510[k] clearance for such agents.

CDC

At CDC, the mission of the Coordinating Center for Infections Diseases is to guide the public on how to prevent and respond to infectious diseases in both health-care settings and at home. With respect to disinfectants and sterilants, part of CDC's role is to inform the public (in this case healthcare personnel) of current scientific evidence pertaining to these products, to comment about their safety and efficacy, and to recommend which chemicals might be most appropriate or effective for specific microorganisms and settings.

Test Methods

The methods EPA has used for registration are standardized by the AOAC International; however, a survey of scientific literature reveals a number of problems with these tests that were reported during 1987–1990 [58, 76, 80, 428, 736, 737, 795-800] that cause them to be neither accurate nor reproducible [416, 737]. As part of their regulatory authority, EPA and FDA support development and validation of methods for assessing disinfection claims [801-803]. For example, EPA has supported the work of Dr. Syed Sattar and coworkers who have developed a two-tier quantitative carrier test to assess sporicidal, mycobactericidal, bactericidal, fungicidal, virucidal, and protozoacidal activity of chemical germicides [701, 803]. EPA is accepting label claims against hepatitis B virus (HBV) using a surrogate organism, the duck HBV, to quantify disinfectant activity [124, 804]. EPA also is accepting labeling claims against hepatitis C virus using the bovine viral diarrhea virus as a surrogate.

For nearly 30 years, EPA also performed intramural preregistration and postregistration efficacy testing of some chemical disinfectants in its own laboratories. In 1982, this was stopped, reportedly for budgetary reasons. At that time, manufacturers did not need to have microbiologic activity claims verified by EPA or an independent testing laboratory when registering a disinfectant or chemical sterilant [805]. This occurred when the frequency of contaminated germicides and infections secondary to their use had increased [404]. Investigations demonstrating that interlaboratory reproducibility of test results was poor and manufacturers' label claims were not verifiable [416, 737] and symposia sponsored by the American Society for Microbiology [800] heightened awareness of these problems and reconfirmed the need to improve the AOAC methods and reinstate a microbiologic activity verification program. A General Accounting Office report entitled *Disinfectants: EPA Lacks Assurance They Work* [806] seemed to provide the necessary impetus for EPA to initiate corrective measures, including cooperative agreements to improve the AOAC methods and independent verification testing for all products labeled as sporicidal and disinfectants labeled as tuberculocidal. For example, of 26 sterilant products tested by EPA, 15 were canceled because of product failure. A list of products registered with EPA and labeled for use as sterilants or tuberculocides or against HIV and/or HBV is available through EPA's website at http://www.epa.gov/oppad001/chemregindex.htm. Organizations (e.g., Organization for Economic Cooperation and Development) are working to standardize requirements for germicide testing and registration.

Neutralization of Germicides

One of the difficulties associated with evaluating the bactericidal activity of disinfectants is prevention of bacteriostasis from disinfectant residues carried over into the subculture media. Likewise, small amounts of disinfectants on environmental surfaces can make an accurate bacterial count difficult to get when sampling of the health-care environment as part of an epidemiologic or research investigation. One way these problems may be overcome is by employing neutralizers that inactivate residual disinfectants [807-809]. Two commonly used neutralizing media for chemical disinfectants are Letheen Media and D/E Neutralizing Media. The former contains lecithin to neutralize quaternaries and polysorbate 80 (Tween 80) to neutralize phenolics, hexachlorophene, formalin, and, with lecithin, ethanol. The D/E Neutralizing media will neutralize a broad spectrum of antiseptic and disinfectant chemicals, including quaternary ammonium compounds, phenols, iodine and chlorine compounds, mercurials, formaldehyde, and glutaraldehyde [810]. A review of neutralizers used in germicide testing has been published[808].

STERILIZATION

Most medical and surgical devices used in healthcare facilities are made of materials that are heat stable and therefore undergo heat, primarily steam, sterilization. However, since 1950, there has been an increase in medical devices and instruments made of materials (e.g., plastics) that require low-temperature sterilization. Ethylene oxide gas has been used since the 1950s for heat- and moisture-sensitive medical devices. Within the past 15 years, a number of new, low-temperature sterilization systems (e.g., hydrogen peroxide gas plasma, peracetic acid immersion, ozone) have been developed and are being used to sterilize medical devices. This section reviews sterilization technologies used in healthcare and makes recommendations for their optimum performance in the processing of medical devices [1, 18, 811-820].

Sterilization destroys all microorganisms on the surface of an article or in a fluid to prevent disease transmission associated with the use of that item. While the use of inadequately sterilized critical items represents a high risk of transmitting pathogens, documented transmission of pathogens associated with an inadequately sterilized critical item is exceedingly rare [821, 822]. This is likely due to the wide margin of safety associated with the sterilization processes used in healthcare facilities. The concept of what constitutes "sterile" is measured as a probability of sterility for each item to be sterilized. This probability is commonly referred to as the sterility assurance level (SAL) of the product and is defined as the probability of a single viable microorganism occurring on a product after sterilization. SAL is normally expressed a 10^{-n}. For example, if the probability of a spore surviving were one in one million, the SAL would be 10^{-6} [823, 824]. In short, a SAL is an estimate of lethality of the entire sterilization process and is a conservative calculation. Dual SALs (e.g., 10^{-3} SAL for blood culture tubes, drainage bags; 10^{-6} SAL for scalpels, implants) have been used in the United States for many years and the choice of a 10^{-6} SAL was strictly arbitrary and not associated with any adverse outcomes (e.g., patient infections) [823].

Medical devices that have contact with sterile body tissues or fluids are considered critical items. These items should be sterile when used because any microbial contamination could result in disease transmission. Such items include surgical instruments, biopsy forceps, and implanted medical devices. If these items are heat resistant, the recommended sterilization process is steam sterilization, because it has the largest margin of safety due to its reliability, consistency, and lethality. However, reprocessing heat- and moisture-sensitive items requires use of a low-temperature sterilization technology (e.g., ethylene oxide, hydrogen peroxide gas plasma, peracetic acid) [825]. A summary of the advantages and disadvantages for commonly used sterilization technologies is presented in Table 6.

Steam Sterilization

Overview. Of all the methods available for sterilization, moist heat in the form of saturated steam under pressure is the most widely used and the most dependable. Steam sterilization is nontoxic, inexpensive [826], rapidly microbicidal, sporicidal, and rapidly heats and penetrates fabrics (Table 6) [827]. Like all sterilization processes, steam sterilization has some deleterious effects on some materials, including corrosion and combustion of lubricants associated with dental handpieces[212]; reduction in ability to transmit light associated with laryngoscopes[828]; and increased hardening time (5.6 fold) with plaster-cast [829].

The basic principle of steam sterilization, as accomplished in an autoclave, is to expose each item to direct steam contact at the required temperature and pressure for the specified time. Thus, there are four parameters of steam sterilization: steam, pressure, temperature, and time. The ideal steam for sterilization is dry saturated steam and entrained water (dryness fraction $\geq$97%)[813, 819]. Pressure serves as a means to obtain the high temperatures necessary to quickly kill microorganisms. Specific temperatures must be obtained to ensure the microbicidal activity. The two common steam-sterilizing temperatures are 121°C (250°F) and 132°C (270°F). These temperatures (and other high temperatures) [830] must be maintained for a minimal time to kill microorganisms. Recognized minimum exposure periods for sterilization of wrapped healthcare supplies are 30 minutes at 121°C (250°F) in a gravity displacement

sterilizer or 4 minutes at 132°C (270°C) in a prevacuum sterilizer (Table 7). At constant temperatures, sterilization times vary depending on the type of item (e.g., metal versus rubber, plastic, items with lumens), whether the item is wrapped or unwrapped, and the sterilizer type.

The two basic types of steam sterilizers (autoclaves) are the gravity displacement autoclave and the high-speed prevacuum sterilizer. In the former, steam is admitted at the top or the sides of the sterilizing chamber and, because the steam is lighter than air, forces air out the bottom of the chamber through the drain vent. The gravity displacement autoclaves are primarily used to process laboratory media, water, pharmaceutical products, regulated medical waste, and nonporous articles whose surfaces have direct steam contact. For gravity displacement sterilizers the penetration time into porous items is prolonged because of incomplete air elimination. This point is illustrated with the decontamination of 10 lbs of microbiological waste, which requires at least 45 minutes at 121°C because the entrapped air remaining in a load of waste greatly retards steam permeation and heating efficiency[831, 832]. The high-speed prevacuum sterilizers are similar to the gravity displacement sterilizers except they are fitted with a vacuum pump (or ejector) to ensure air removal from the sterilizing chamber and load before the steam is admitted. The advantage of using a vacuum pump is that there is nearly instantaneous steam penetration even into porous loads. The Bowie-Dick test is used to detect air leaks and inadequate air removal and consists of folded 100% cotton surgical towels that are clean and preconditioned. A commercially available Bowie-Dick-type test sheet should be placed in the center of the pack. The test pack should be placed horizontally in the front, bottom section of the sterilizer rack, near the door and over the drain, in an otherwise empty chamber and run at 134°C for 3.5 minutes[813, 819]. The test is used each day the vacuum-type steam sterilizer is used, before the first processed load. Air that is not removed from the chamber will interfere with steam contact. Smaller disposable test packs (or process challenge devices) have been devised to replace the stack of folded surgical towels for testing the efficacy of the vacuum system in a prevacuum sterilizer. [833] These devices are "designed to simulate product to be sterilized and to constitute a defined challenge to the sterilization process"[819, 834]. They should be representative of the load and simulate the greatest challenge to the load[835]. Sterilizer vacuum performance is acceptable if the sheet inside the test pack shows a uniform color change. Entrapped air will cause a spot to appear on the test sheet, due to the inability of the steam to reach the chemical indicator. If the sterilizer fails the Bowie-Dick test, do not use the sterilizer until it is inspected by the sterilizer maintenance personnel and passes the Bowie-Dick test[813, 819, 836].

Another design in steam sterilization is a steam flush-pressure pulsing process, which removes air rapidly by repeatedly alternating a steam flush and a pressure pulse above atmospheric pressure. Air is rapidly removed from the load as with the prevacuum sterilizer, but air leaks do not affect this process because the steam in the sterilizing chamber is always above atmospheric pressure. Typical sterilization temperatures and times are 132°C to 135°C with 3 to 4 minutes exposure time for porous loads and instruments[827, 837].

Like other sterilization systems, the steam cycle is monitored by mechanical, chemical, and biological monitors. Steam sterilizers usually are monitored using a printout (or graphically) by measuring temperature, the time at the temperature, and pressure. Typically, chemical indicators are affixed to the outside and incorporated into the pack to monitor the temperature or time and temperature. The effectiveness of steam sterilization is monitored with a biological indicator containing spores of *Geobacillus stearothermophilus* (formerly *Bacillus stearothermophilus*). Positive spore test results are a relatively rare event [838] and can be attributed to operator error, inadequate steam delivery[839], or equipment malfunction.

Portable (table-top) steam sterilizers are used in outpatient, dental, and rural clinics[840]. These sterilizers are designed for small instruments, such as hypodermic syringes and needles and dental instruments. The ability of the sterilizer to reach physical parameters necessary to achieve sterilization should be monitored by mechanical, chemical, and biological indicators.

Microbicidal Activity. The oldest and most recognized agent for inactivation of microorganisms is heat. D-values (time to reduce the surviving population by 90% or 1 $\log_{10}$) allow a direct comparison of the heat resistance of microorganisms. Because a D-value can be determined at various temperatures, a subscript is used to designate the exposure temperature (i.e., D_{121C}). D_{121C}-values for *Geobacillus stearothermophilus* used to monitor the steam sterilization process range from 1 to 2 minutes. Heat-resistant nonspore-forming bacteria, yeasts, and fungi have such low D_{121C} values that they cannot be experimentally measured[841].

Mode of Action. Moist heat destroys microorganisms by the irreversible coagulation and denaturation of enzymes and structural proteins. In support of this fact, it has been found that the presence of moisture significantly affects the coagulation temperature of proteins and the temperature at which microorganisms are destroyed.

Uses. Steam sterilization should be used whenever possible on all critical and semicritical items that are heat and moisture resistant (e.g., steam sterilizable respiratory therapy and anesthesia equipment), even when not essential to prevent pathogen transmission. Steam sterilizers also are used in healthcare facilities to decontaminate microbiological waste and sharps containers [831, 832, 842] but additional exposure time is required in the gravity displacement sterilizer for these items.

Flash Sterilization

Overview. "Flash" steam sterilization was originally defined by Underwood and Perkins as sterilization of an unwrapped object at 132°C for 3 minutes at 27-28 lbs. of pressure in a gravity displacement sterilizer[843]. Currently, the time required for flash sterilization depends on the type of sterilizer and the type of item (i.e., porous vs non-porous items)(see Table 8). Although the wrapped method of sterilization is preferred for the reasons listed below, correctly performed flash sterilization is an effective process for the sterilization of critical medical devices[844, 845]. Flash sterilization is a modification of conventional steam sterilization (either gravity, prevacuum, or steam-flush pressure-pulse) in which the flashed item is placed in an open tray or is placed in a specially designed, covered, rigid container to allow for rapid penetration of steam. Historically, it is not recommended as a routine sterilization method because of the lack of timely biological indicators to monitor performance, absence of protective packaging following sterilization, possibility for contamination of processed items during transportation to the operating rooms, and the sterilization cycle parameters (i.e., time, temperature, pressure) are minimal. To address some of these concerns, many healthcare facilities have done the following: placed equipment for flash sterilization in close proximity to operating rooms to facilitate aseptic delivery to the point of use (usually the sterile field in an ongoing surgical procedure); extended the exposure time to ensure lethality comparable to sterilized wrapped items (e.g., 4 minutes at 132°C)[846, 847]; used biological indicators that provide results in 1 hour for flash-sterilized items[846, 847]; and used protective packaging that permits steam penetration[812, 817-819, 845, 848]. Further, some rigid, reusable sterilization container systems have been designed and validated by the container manufacturer for use with flash cycles. When sterile items are open to air, they will eventually become contaminated. Thus, the longer a sterile item is exposed to air, the greater the number of microorganisms that will settle on it. Sterilization cycle parameters for flash sterilization are shown in Table 8.

A few adverse events have been associated with flash sterilization. When evaluating an increased incidence of neurosurgical infections, the investigators noted that surgical instruments were flash sterilized between cases and 2 of 3 craniotomy infections involved plate implants that were flash sterilized[849]. A report of two patients who received burns during surgery from instruments that had been flash sterilized reinforced the need to develop policies and educate staff to prevent the use of instruments hot enough to cause clinical burns[850]. Staff should use precautions to prevent burns with potentially hot instruments (e.g., transport tray using heat-protective gloves). Patient burns may be prevented by either air-cooling the instruments or immersion in sterile liquid (e.g., saline).

Uses. Flash sterilization is considered acceptable for processing cleaned patient-care items that

cannot be packaged, sterilized, and stored before use. It also is used when there is insufficient time to sterilize an item by the preferred package method. Flash sterilization should not be used for reasons of convenience, as an alternative to purchasing additional instrument sets, or to save time[817]. Because of the potential for serious infections, flash sterilization is not recommended for implantable devices (i.e., devices placed into a surgically or naturally formed cavity of the human body); however, flash sterilization may be unavoidable for some devices (e.g., orthopedic screw, plates). If flash sterilization of an implantable device is unavoidable, recordkeeping (i.e., load identification, patient's name/hospital identifier, and biological indicator result) is essential for epidemiological tracking (e.g., of surgical site infection, tracing results of biological indicators to patients who received the item to document sterility), and for an assessment of the reliability of the sterilization process (e.g., evaluation of biological monitoring records and sterilization maintenance records noting preventive maintenance and repairs with dates).

Low-Temperature Sterilization Technologies

Ethylene oxide (ETO) has been widely used as a low-temperature sterilant since the 1950s. It has been the most commonly used process for sterilizing temperature- and moisture-sensitive medical devices and supplies in healthcare institutions in the United States. Two types of ETO sterilizers are available, mixed gas and 100% ETO. Until 1995, ethylene oxide sterilizers combined ETO with a chloroflourocarbon (CFC) stabilizing agent, most commonly in a ratio of 12% ETO mixed with 88% CFC (referred to as 12/88 ETO).

For several reasons, healthcare personnel have been exploring the use of new low-temperature sterilization technologies[825, 851]. First, CFCs were phased out in December 1995 under provisions of the Clean Air Act [852]. CFCs were classified as a Class I substance under the Clean Air Act because of scientific evidence linking them to destruction of the earth's ozone layer. Second, some states (e.g., California, New York, Michigan) require the use of ETO abatement technology to reduce the amount of ETO being released into ambient air from 90 to 99.9% depending on the state. Third, OSHA regulates the acceptable vapor levels of ETO (i.e., 1 ppm averaged over 8 hours) due to concerns that ETO exposure represents an occupational hazard[318]. These constraints have led to the development of alternative technologies for low-temperature sterilization in the healthcare setting.

Alternative technologies to ETO with chlorofluorocarbon that are currently available and cleared by the FDA for medical equipment include 100% ETO; ETO with a different stabilizing gas, such as carbon dioxide or hydrochlorofluorocarbons (HCFC); immersion in peracetic acid; hydrogen peroxide gas plasma; and ozone. Technologies under development for use in healthcare facilities, but not cleared by the FDA, include vaporized hydrogen peroxide, vapor phase peracetic acid, gaseous chlorine dioxide, ionizing radiation, or pulsed light [400, 758, 853]. However, there is no guarantee that these new sterilization technologies will receive FDA clearance for use in healthcare facilities.

These new technologies should be compared against the characteristics of an ideal low-temperature (<60^{o}C) sterilant (Table 9). [851] While it is apparent that all technologies will have limitations (Table 9), understanding the limitations imposed by restrictive device designs (e.g., long, narrow lumens) is critical for proper application of new sterilization technology[854]. For example, the development of increasingly small and complex endoscopes presents a difficult challenge for current sterilization processes. This occurs because microorganisms must be in direct contact with the sterilant for inactivation to occur. Several peer-reviewed scientific publications have data demonstrating concerns about the efficacy of several of the low-temperature sterilization processes (i.e., gas plasma, vaporized hydrogen peroxide, ETO, peracetic acid), particularly when the test organisms are challenged in the presence of serum and salt and a narrow lumen vehicle[469, 721, 825, 855, 856]. Factors shown to affect the efficacy of sterilization are shown in Table 10.

Ethylene Oxide "Gas" Sterilization

Overview. ETO is a colorless gas that is flammable and explosive. The four essential

parameters (operational ranges) are: gas concentration (450 to 1200 mg/l); temperature (37 to 63°C); relative humidity (40 to 80%)(water molecules carry ETO to reactive sites); and exposure time (1 to 6 hours). These influence the effectiveness of ETO sterilization[814, 857, 858]. Within certain limitations, an increase in gas concentration and temperature may shorten the time necessary for achieving sterilization.

The main disadvantages associated with ETO are the lengthy cycle time, the cost, and its potential hazards to patients and staff; the main advantage is that it can sterilize heat- or moisture-sensitive medical equipment without deleterious effects on the material used in the medical devices (Table 6). Acute exposure to ETO may result in irritation (e.g., to skin, eyes, gastrointestinal or respiratory tracts) and central nervous system depression[859-862]. Chronic inhalation has been linked to the formation of cataracts, cognitive impairment, neurologic dysfunction, and disabling polyneuropathies[860, 861, 863-866]. Occupational exposure in healthcare facilities has been linked to hematologic changes [867] and an increased risk of spontaneous abortions and various cancers[318, 868-870]. ETO should be considered a known human carcinogen[871].

The basic ETO sterilization cycle consists of five stages (i.e., preconditioning and humidification, gas introduction, exposure, evacuation, and air washes) and takes approximately 2 1/2 hrs excluding aeration time. Mechanical aeration for 8 to 12 hours at 50 to 60°C allows desorption of the toxic ETO residual contained in exposed absorbent materials. Most modern ETO sterilizers combine sterilization and aeration in the same chamber as a continuous process. These ETO models minimize potential ETO exposure during door opening and load transfer to the aerator. Ambient room aeration also will achieve desorption of the toxic ETO but requires 7 days at 20°C. There are no federal regulations for ETO sterilizer emission; however, many states have promulgated emission-control regulations[814].

The use of ETO evolved when few alternatives existed for sterilizing heat- and moisture-sensitive medical devices; however, favorable properties (Table 6) account for its continued widespread use[872]. Two ETO gas mixtures are available to replace ETO-chlorofluorocarbon (CFC) mixtures for large capacity, tank-supplied sterilizers. The ETO-carbon dioxide (CO_2) mixture consists of 8.5% ETO and 91.5% CO_2. This mixture is less expensive than ETO-hydrochlorofluorocarbons (HCFC), but a disadvantage is the need for pressure vessels rated for steam sterilization, because higher pressures (28-psi gauge) are required. The other mixture, which is a drop-in CFC replacement, is ETO mixed with HCFC. HCFCs are approximately 50-fold less damaging to the earth's ozone layer than are CFCs. The EPA will begin regulation of HCFC in the year 2015 and will terminate production in the year 2030. Two companies provide ETO-HCFC mixtures as drop-in replacement for CFC-12; one mixture consists of 8.6% ETO and 91.4% HCFC, and the other mixture is composed of 10% ETO and 90% HCFC[872]. An alternative to the pressurized mixed gas ETO systems is 100% ETO. The 100% ETO sterilizers using unit-dose cartridges eliminate the need for external tanks.

ETO is absorbed by many materials. For this reason, following sterilization the item must undergo aeration to remove residual ETO. Guidelines have been promulgated regarding allowable ETO limits for devices that depend on how the device is used, how often, and how long in order to pose a minimal risk to patients in normal product use[814].

ETO toxicity has been established in a variety of animals. Exposure to ETO can cause eye pain, sore throat, difficulty breathing and blurred vision. Exposure can also cause dizziness, nausea, headache, convulsions, blisters and vomiting and coughing[873]. In a variety of *in vitro* and animal studies, ETO has been demonstrated to be carcinogenic. ETO has been linked to spontaneous abortion, genetic damage, nerve damage, peripheral paralysis, muscle weakness, and impaired thinking and memory[873]. Occupational exposure in healthcare facilities has been linked to an increased risk of spontaneous abortions and various cancers[318]. Injuries (e.g., tissue burns) to patients have been associated with ETO residues in implants used in surgical procedures[874]. Residual ETO in capillary flow dialysis membranes has been shown to be neurotoxic in vitro[875]. OSHA has established a PEL of 1 ppm airborne ETO in the workplace, expressed as a TWA for an 8-hour work shift in a 40-hour work week. The "action level" for ETO is 0.5 ppm, expressed as an 8-hour TWA, and the short-term excursion limit is 5 ppm, expressed as

a 15-minute TWA[814]. For details of the requirements in OSHA's ETO standard for occupational exposures, see Title 29 of the Code of Federal Regulations (CFR) Part 1910.1047[873]. Several personnel monitoring methods (e.g., charcoal tubes and passive sampling devices) are in use[814]. OSHA has established a PEL of 5 ppm for ethylene chlorohydrin (a toxic by-product of ETO) in the workplace[876]. Additional information regarding use of ETO in health care facilities is available from NIOSH.

Mode of Action. The microbicidal activity of ETO is considered to be the result of alkylation of protein, DNA, and RNA. Alkylation, or the replacement of a hydrogen atom with an alkyl group, within cells prevents normal cellular metabolism and replication[877].

Microbicidal Activity. The excellent microbicidal activity of ETO has been demonstrated in several studies [469, 721, 722, 856, 878, 879] and summarized in published reports[877]. ETO inactivates all microorganisms although bacterial spores (especially *B. atrophaeus*) are more resistant than other microorganisms. For this reason *B. atrophaeus* is the recommended biological indicator.

Like all sterilization processes, the effectiveness of ETO sterilization can be altered by lumen length, lumen diameter, inorganic salts, and organic materials[469, 721, 722, 855, 856, 879]. For example, although ETO is not used commonly for reprocessing endoscopes[28], several studies have shown failure of ETO in inactivating contaminating spores in endoscope channels [855]or lumen test units [469, 721, 879] and residual ETO levels averaging 66.2 ppm even after the standard degassing time[456]. Failure of ETO also has been observed when dental handpieces were contaminated with *Streptococcus mutans* and exposed to ETO[880]. It is recommended that dental handpieces be steam sterilized.

Uses. ETO is used in healthcare facilities to sterilize critical items (and sometimes semicritical items) that are moisture or heat sensitive and cannot be sterilized by steam sterilization.

Hydrogen Peroxide Gas Plasma

Overview. New sterilization technology based on plasma was patented in 1987 and marketed in the United States in 1993. Gas plasmas have been referred to as the fourth state of matter (i.e., liquids, solids, gases, and gas plasmas). Gas plasmas are generated in an enclosed chamber under deep vacuum using radio frequency or microwave energy to excite the gas molecules and produce charged particles, many of which are in the form of free radicals. A free radical is an atom with an unpaired electron and is a highly reactive species. The proposed mechanism of action of this device is the production of free radicals within a plasma field that are capable of interacting with essential cell components (e.g., enzymes, nucleic acids) and thereby disrupt the metabolism of microorganisms. The type of seed gas used and the depth of the vacuum are two important variables that can determine the effectiveness of this process.

In the late 1980s the first hydrogen peroxide gas plasma system for sterilization of medical and surgical devices was field-tested. According to the manufacturer, the sterilization chamber is evacuated and hydrogen peroxide solution is injected from a cassette and is vaporized in the sterilization chamber to a concentration of 6 mg/l. The hydrogen peroxide vapor diffuses through the chamber (50 minutes), exposes all surfaces of the load to the sterilant, and initiates the inactivation of microorganisms. An electrical field created by a radio frequency is applied to the chamber to create a gas plasma. Microbicidal free radicals (e.g., hydroxyl and hydroperoxyl) are generated in the plasma. The excess gas is removed and in the final stage (i.e., vent) of the process the sterilization chamber is returned to atmospheric pressure by introduction of high-efficiency filtered air. The by-products of the cycle (e.g., water vapor, oxygen) are nontoxic and eliminate the need for aeration. Thus, the sterilized materials can be handled safely, either for immediate use or storage. The process operates in the range of 37-44°C and has a cycle time of 75 minutes. If any moisture is present on the objects the vacuum will not be achieved and the cycle aborts[856, 881-883].

A newer version of the unit improves sterilizer efficacy by using two cycles with a hydrogen

peroxide diffusion stage and a plasma stage per sterilization cycle. This revision, which is achieved by a software modification, reduces total processing time from 73 to 52 minutes. The manufacturer believes that the enhanced activity obtained with this system is due in part to the pressure changes that occur during the injection and diffusion phases of the process and to the fact that the process consists of two equal and consecutive half cycles, each with a separate injection of hydrogen peroxide. [856, 884, 885] This system and a smaller version [400, 882] have received FDA 510[k] clearance with limited application for sterilization of medical devices (Table 6). The biological indicator used with this system is *Bacillus atrophaeus* spores[851]. The newest version of the unit, which employs a new vaporization system that removes most of the water from the hydrogen peroxide, has a cycle time from 28-38 minutes (see manufacturer's literature for device dimension restrictions).

Penetration of hydrogen peroxide vapor into long or narrow lumens has been addressed outside the United States by the use of a diffusion enhancer. This is a small, breakable glass ampoule of concentrated hydrogen peroxide (50%) with an elastic connector that is inserted into the device lumen and crushed immediately before sterilization[470, 885]. The diffusion enhancer has been shown to sterilize bronchoscopes contaminated with *Mycobacteria tuberculosis*[886]. At the present time, the diffusion enhancer is not FDA cleared.

Another gas plasma system, which differs from the above in several important ways, including the use of peracetic acid-acetic acid-hydrogen peroxide vapor, was removed from the marketplace because of reports of corneal destruction to patients when ophthalmic surgery instruments had been processed in the sterilizer[887, 888]. In this investigation, exposure of potentially wet ophthalmologic surgical instruments with small bores and brass components to the plasma gas led to degradation of the brass to copper and zinc[888, 889]. The experimenters showed that when rabbit eyes were exposed to the rinsates of the gas plasma-sterilized instruments, corneal decompensation was documented. This toxicity is highly unlikely with the hydrogen peroxide gas plasma process since a toxic, soluble form of copper would not form (LA Feldman, written communication, April 1998).

Mode of Action. This process inactivates microorganisms primarily by the combined use of hydrogen peroxide gas and the generation of free radicals (hydroxyl and hydroproxyl free radicals) during the plasma phase of the cycle.

***Microbicidal Activity*.** This process has the ability to inactivate a broad range of microorganisms, including resistant bacterial spores. Studies have been conducted against vegetative bacteria (including mycobacteria), yeasts, fungi, viruses, and bacterial spores[469, 721, 856, 881-883, 890-893]. Like all sterilization processes, the effectiveness can be altered by lumen length, lumen diameter, inorganic salts, and organic materials[469, 721, 855, 856, 890, 891, 893].

Uses. Materials and devices that cannot tolerate high temperatures and humidity, such as some plastics, electrical devices, and corrosion-susceptible metal alloys, can be sterilized by hydrogen peroxide gas plasma. This method has been compatible with most (>95%) medical devices and materials tested[884, 894, 895].

Peracetic Acid Sterilization

Overview. Peracetic acid is a highly biocidal oxidizer that maintains its efficacy in the presence of organic soil. Peracetic acid removes surface contaminants (primarily protein) on endoscopic tubing[711, 717]. An automated machine using peracetic acid to sterilize medical, surgical, and dental instruments chemically (e.g., endoscopes, arthroscopes) was introduced in 1988. This microprocessor-controlled, low-temperature sterilization method is commonly used in the United States[107]. The sterilant, 35% peracetic acid, and an anticorrosive agent are supplied in a single-dose container. The container is punctured at the time of use, immediately prior to closing the lid and initiating the cycle. The concentrated peracetic acid is diluted to 0.2% with filtered water (0.2 μm) at a temperature of approximately 50°C. The diluted peracetic acid is circulated within the chamber of the machine and

pumped through the channels of the endoscope for 12 minutes, decontaminating exterior surfaces, lumens, and accessories. Interchangeable trays are available to permit the processing of up to three rigid endoscopes or one flexible endoscope. Connectors are available for most types of flexible endoscopes for the irrigation of all channels by directed flow. Rigid endoscopes are placed within a lidded container, and the sterilant fills the lumens either by immersion in the circulating sterilant or by use of channel connectors to direct flow into the lumen(s) (see below for the importance of channel connectors). The peracetic acid is discarded via the sewer and the instrument rinsed four times with filtered water. Concern has been raised that filtered water may be inadequate to maintain sterility[896]. Limited data have shown that low-level bacterial contamination may follow the use of filtered water in an AER but no data has been published on AERs using the peracetic acid system[161]. Clean filtered air is passed through the chamber of the machine and endoscope channels to remove excess water[719]. As with any sterilization process, the system can only sterilize surfaces that can be contacted by the sterilant. For example, bronchoscopy-related infections occurred when bronchoscopes were processed using the wrong connector[155, 725]. Investigation of these incidents revealed that bronchoscopes were inadequately reprocessed when inappropriate channel connectors were used and when there were inconsistencies between the reprocessing instructions provided by the manufacturer of the bronchoscope and the manufacturer of the automatic endoscope reprocessor[155]. The importance of channel connectors to achieve sterilization was also shown for rigid lumen devices[137, 856].

The manufacturers suggest the use of biological monitors (*G. stearothermophilus* spore strips) both at the time of installation and routinely to ensure effectiveness of the process. The manufacturer's clip must be used to hold the strip in the designated spot in the machine as a broader clamp will not allow the sterilant to reach the spores trapped under it[897]. One investigator reported a 3% failure rate when the appropriate clips were used to hold the spore strip within the machine[718]. The use of biological monitors designed to monitor either steam sterilization or ETO for a liquid chemical sterilizer has been questioned for several reasons including spore wash-off from the filter paper strips which may cause less valid monitoring[898-901]. The processor is equipped with a conductivity probe that will automatically abort the cycle if the buffer system is not detected in a fresh container of the peracetic acid solution. A chemical monitoring strip that detects that the active ingredient is >1500 ppm is available for routine use as an additional process control.

Mode of Action. Only limited information is available regarding the mechanism of action of peracetic acid, but it is thought to function as other oxidizing agents, i.e., it denatures proteins, disrupts cell wall permeability, and oxidizes sulfhydral and sulfur bonds in proteins, enzymes, and other metabolites[654, 726].

Microbicidal Activity. Peracetic acid will inactivate gram-positive and gram-negative bacteria, fungi, and yeasts in <5 minutes at <100 ppm. In the presence of organic matter, 200-500 ppm is required. For viruses, the dosage range is wide (12-2250 ppm), with poliovirus inactivated in yeast extract in 15 minutes with 1500 to 2250 ppm. Bacterial spores in suspension are inactivated in 15 seconds to 30 minutes with 500 to 10,000 ppm (0.05 to 1%)[654].

Simulated-use trials have demonstrated microbicidal activity [111, 718-722] and three clinical trials have demonstrated both microbial killing and no clinical failures leading to infection[90, 723, 724]. Alfa and co-workers, who compared the peracetic acid system with ETO, demonstrated the high efficacy of the system. Only the peracetic acid system was able to completely kill 6-$\log_{10}$ of *Mycobacterium chelonae*, *Enterococcus faecalis*, and *B. atrophaeus* spores with both an organic and inorganic challenge[722]. Like other sterilization processes, the efficacy of the process can be diminished by soil challenges [902] and test conditions[856].

Uses. This automated machine is used to chemically sterilize medical (e.g., GI endoscopes) and surgical (e.g., flexible endoscopes) instruments in the United States. Lumened endoscopes must be connected to an appropriate channel connector to ensure that the sterilant has direct contact with the contaminated lumen. [137, 856, 903] Olympus America has not listed this system as a compatible product for

use in reprocessing Olympus bronchoscopes and gastrointestinal endoscopes (Olympus America, January 30, 2002, written communication).

Microbicidal Activity of Low-Temperature Sterilization Technologies

Sterilization processes used in the United States must be cleared by FDA, and they require that sterilizer microbicidal performance be tested under simulated-use conditions[904]. FDA requires that the test article be inoculated with 10^6 colony-forming units of the most resistant test organism and prepared with organic and inorganic test loads as would occur after actual use. FDA requires manufacturers to use organic soil (e.g., 5% fetal calf serum), dried onto the device with the inoculum, to represent soil remaining on the device following marginal cleaning. However, 5% fetal calf serum as a measure of marginal cleaning has not been validated by measurements of protein load on devices following use and the level of protein removal by various cleaning methods. The inocula must be placed in various locations of the test articles, including those least favorable to penetration and contact with the sterilant (e.g., lumens). Cleaning before sterilization is not allowed in the demonstration of sterilization efficacy[904]. Several studies have evaluated the relative microbicidal efficacy of these low-temperature sterilization technologies (Table 11). These studies have either tested the activity of a sterilization process against specific microorganisms[892, 905, 906], evaluated the microbicidal activity of a singular technology [711, 719, 724, 855, 879, 882-884, 890, 891, 907] or evaluated the comparative effectiveness of several sterilization technologies[271, 426, 469, 721, 722, 856, 908, 909]. Several test methodologies use stainless steel or porcelain carriers that are inoculated with a test organism. Commonly used test organisms include vegetative bacteria, mycobacteria, and spores of *Bacillus* species. The available data demonstrate that low-temperature sterilization technologies are able to provide a 6-$\log_{10}$ reduction of microbes when inoculated onto carriers in the absence of salt and serum. However, tests can be constructed such that all of the available sterilization technologies are unable to reliably achieve complete inactivation of a microbial load.[425, 426, 469, 721, 856, 909] For example, almost all of the sterilization processes will fail to reliably inactivate the microbial load in the presence of salt and serum[469, 721, 909].

The effect of salts and serums on the sterilization process were studied initially in the 1950s and 1960s[424, 910]. These studies showed that a high concentration of crystalline-type materials and a low protein content provided greater protection to spores than did serum with a high protein content[426]. A study by Doyle and Ernst demonstrated resistance of spores by crystalline material applied not only to low-temperature sterilization technology but also to steam and dry heat[425]. These studies showed that occlusion of *Bacillus atrophaeus* spores in calcium carbonate crystals dramatically increased the time required for inactivation as follows: 10 seconds to 150 minutes for steam (121°C), 3.5 hours to 50 hours for dry heat (121°C), 30 seconds to >2 weeks for ETO (54°C). Investigators have corroborated and extended these findings[469, 470, 721, 855, 908, 909]. While soils containing both organic and inorganic materials impair microbial killing, soils that contain a high inorganic salt-to-protein ratio favor crystal formation and impair sterilization by occlusion of organisms[425, 426, 881].

Alfa and colleagues demonstrated a 6-$\log_{10}$ reduction of the microbial inoculum of porcelain penicylinders using a variety of vegetative and spore-forming organisms (Table 11)[469]. However, if the bacterial inoculum was in tissue-culture medium supplemented with 10% serum, only the ETO 12/88 and ETO-HCFC sterilization mixtures could sterilize 95% to 97% of the penicylinder carriers. The plasma and 100% ETO sterilizer demonstrated significantly reduced activity (Table 11). For all sterilizers evaluated using penicylinder carriers (i.e., ETO 12/88, 100% ETO, hydrogen peroxide gas plasma), there was a 3- to 6-$\log_{10}$ reduction of inoculated bacteria even in the presence of serum and salt. For each sterilizer evaluated, the ability to inactivate microorganisms in the presence of salt and serum was reduced even further when the inoculum was placed in a narrow-lumen test object (3 mm diameter by 125 cm long). Although there was a 2- to 4-$\log_{10}$ reduction in microbial kill, less than 50% of the lumen test objects were sterile when processed using any of the sterilization methods evaluated except the peracetic acid immersion system (Table 11)[721]. Complete killing (or removal) of 6-$\log_{10}$ of *Enterococcus faecalis, Mycobacterium chelonei*, and *Bacillus atrophaeus* spores in the presence of salt and serum and lumen test objects was observed only for the peracetic acid immersion system.

With respect to the results by Alfa and coworkers[469], Jacobs showed that the use of the tissue culture media created a technique-induced sterilization failure[426]. Jacobs et al. showed that microorganisms mixed with tissue culture media, used as a surrogate body fluid, formed physical crystals that protected the microorganisms used as a challenge. If the carriers were exposed for 60 sec to nonflowing water, the salts dissolved and the protective effect disappeared. Since any device would be exposed to water for a short period of time during the washing procedure, these protective effects would have little clinical relevance[426].

Narrow lumens provide a challenge to some low-temperature sterilization processes. For example, Rutala and colleagues showed that, as lumen size decreased, increased failures occurred with some low-temperature sterilization technologies. However, some low-temperature processes such as ETO-HCFC and the hydrogen peroxide gas plasma process remained effective even when challenged by a lumen as small as 1 mm in the absence of salt and serum[856].

The importance of allowing the sterilant to come into contact with the inoculated carrier is demonstrated by comparing the results of two investigators who studied the peracetic acid immersion system. Alfa and coworkers demonstrated excellent activity of the peracetic acid immersion system against three test organisms using a narrow-lumen device. In these experiments, the lumen test object was connected to channel irrigators, which ensured that the sterilant had direct contact with the contaminated carriers[722]. This effectiveness was achieved through a combination of organism wash-off and peracetic acid sterilant killing the test organisms[722]. The data reported by Rutala et al. demonstrated failure of the peracetic acid immersion system to eliminate *Geobacillus stearothermophilus* spores from a carrier placed in a lumen test object. In these experiments, the lumen test unit was not connected to channel irrigators. The authors attributed the failure of the peracetic acid immersion system to eliminate the high levels of spores from the center of the test unit to the inability of the peracetic acid to diffuse into the center of 40-cm long, 3-mm diameter tubes. This may be caused by an air lock or air bubbles formed in the lumen, impeding the flow of the sterilant through the long and narrow lumen and limiting complete access to the *Bacillus* spores[137, 856]. Experiments using a channel connector specifically designed for 1-, 2-, and 3-mm lumen test units with the peracetic acid immersion system were completely effective in eliminating an inoculum of 10^6 *Geobacillus stearothermophilus* spores[7]. The restricted diffusion environment that exists in the test conditions would not exist with flexible scopes processed in the peracetic acid immersion system, because the scopes are connected to channel irrigators to ensure that the sterilant has direct contact with contaminated surfaces. Alfa and associates attributed the efficacy of the peracetic acid immersion system to the ability of the liquid chemical process to dissolve salts and remove protein and bacteria due to the flushing action of the fluid[722].

Bioburden of Surgical Devices

In general, used medical devices are contaminated with a relatively low bioburden of organisms[179, 911, 912]. Nystrom evaluated medical instruments used in general surgical, gynecological, orthopedic, and ear-nose-throat operations and found that 62% of the instruments were contaminated with $<10^1$ organisms after use, 82% with $<10^2$, and 91% with $<10^3$. After being washed in an instrument washer, more than 98% of the instruments had $<10^1$ organisms, and none $>10^2$ organisms[911]. Other investigators have published similar findings[179, 912]. For example, after a standard cleaning procedure, 72% of 50 surgical instruments contained $<10^1$ organisms, 86% $<10^2$, and only 6% had $>3 \times 10^2$[912]. In another study of rigid-lumen medical devices, the bioburden on both the inner and outer surface of the lumen ranged from 10^1 to 10^4 organisms per device. After cleaning, 83% of the devices had a bioburden $\leq 10^2$ organisms[179]. In all of these studies, the contaminating microflora consisted mainly of vegetative bacteria, usually of low pathogenicity (e.g., coagulase-negative *Staphylococcus*)[179, 911, 912].

An evaluation of the microbial load on used critical medical devices such as spinal anesthesia needles and angiographic catheters and sheaths demonstrated that mesophilic microorganisms were detected at levels of 10^1 to 10^2 in only two of five needles. The bioburden on used angiographic

catheters and sheath introducers exceeded 10^3 CFUs on 14% (3 of 21) and 21% (6 of 28), respectively[907].

Effect of Cleaning on Sterilization Efficacy

The effect of salt and serum on the efficacy of low-temperature sterilization technologies has raised concern regarding the margin of safety of these technologies. Experiments have shown that salts have the greatest impact on protecting microorganisms from killing[426, 469]. However, other studies have suggested that these concerns may not be clinically relevant. One study evaluated the relative rate of removal of inorganic salts, organic soil, and microorganisms from medical devices to better understand the dynamics of the cleaning process[426]. These tests were conducted by inoculating Alfa soil (tissue-culture media and 10% fetal bovine serum) [469] containing 10^6 *G. stearothermophilus* spores onto the surface of a stainless-steel scalpel blade. After drying for 30 minutes at 35°C followed by 30 minutes at room temperature, the samples were placed in water at room temperature. The blades were removed at specified times, and the concentration of total protein and chloride ion was measured. The results showed that soaking in deionized water for 60 seconds resulted in a >95% release rate of chloride ion from NaCl solution in 20 seconds, Alfa soil in 30 seconds, and fetal bovine serum in 120 seconds. Thus, contact with water for short periods, even in the presence of protein, rapidly leads to dissolution of salt crystals and complete inactivation of spores by a low-temperature sterilization process (Table 10). Based on these experimental data, cleaning procedures would eliminate the detrimental effect of high salt content on a low-temperature sterilization process.

These articles [426, 469, 721] assessing low-temperature sterilization technology reinforce the importance of meticulous cleaning before sterilization. These data support the critical need for healthcare facilities to develop rigid protocols for cleaning contaminated objects before sterilization[472]. Sterilization of instruments and medical devices is compromised if the process is not preceded by meticulous cleaning.

The cleaning of any narrow-lumen medical device used in patient care presents a major challenge to reprocessing areas. While attention has been focused on flexible endoscopes, cleaning issues related to other narrow-lumen medical devices such as sphinctertomes have been investigated[913]. This study compared manual cleaning with that of automated cleaning with a narrow-lumen cleaner and found that only retro-flushing with the narrow lumen cleaner provided adequate cleaning of the three channels. If reprocessing was delayed for more than 24 hours, retro-flush cleaning was no longer effective and ETO sterilization failure was detected when devices were held for 7 days [913]. In another study involving simulated-use cleaning of laparoscopic devices, Alfa found that minimally the use of retro-flushing should be used during cleaning of non-ported laparoscopic devices[914].

Other Sterilization Methods

Ionizing Radiation. Sterilization by ionizing radiation, primarily by cobalt 60 gamma rays or electron accelerators, is a low-temperature sterilization method that has been used for a number of medical products (e.g., tissue for transplantation, pharmaceuticals, medical devices). There are no FDA-cleared ionizing radiation sterilization processes for use in healthcare facilities. Because of high sterilization costs, this method is an unfavorable alternative to ETO and plasma sterilization in healthcare facilities but is suitable for large-scale sterilization. Some deleterious effects on patient-care equipment associated with gamma radiation include induced oxidation in polyethylene [915] and delamination and cracking in polyethylene knee bearings[916]. Several reviews [917, 918] dealing with the sources, effects, and application of ionizing radiation may be referred to for more detail.

***Dry-Heat Sterilizers*.** This method should be used only for materials that might be damaged by moist heat or that are impenetrable to moist heat (e.g., powders, petroleum products, sharp instruments). The advantages for dry heat include the following: it is nontoxic and does not harm the environment; a dry heat cabinet is easy to install and has relatively low operating costs; it penetrates materials; and it is noncorrosive for metal and sharp instruments. The disadvantages for dry heat are the slow rate of heat penetration and microbial killing makes this a time-consuming method. In addition, the high temperatures

are not suitable for most materials[919]. The most common time-temperature relationships for sterilization with hot air sterilizers are 170°C (340°F) for 60 minutes, 160°C (320°F) for 120 minutes, and 150°C (300°F) for 150 minutes. *B. atrophaeus* spores should be used to monitor the sterilization process for dry heat because they are more resistant to dry heat than are *G. stearothermophilus* spores. The primary lethal process is considered to be oxidation of cell constituents.

There are two types of dry-heat sterilizers: the static-air type and the forced-air type. The static-air type is referred to as the oven-type sterilizer as heating coils in the bottom of the unit cause the hot air to rise inside the chamber via gravity convection. This type of dry-heat sterilizer is much slower in heating, requires longer time to reach sterilizing temperature, and is less uniform in temperature control throughout the chamber than is the forced-air type. The forced-air or mechanical convection sterilizer is equipped with a motor-driven blower that circulates heated air throughout the chamber at a high velocity, permitting a more rapid transfer of energy from the air to the instruments[920].

Liquid Chemicals. Several FDA-cleared liquid chemical sterilants include indications for sterilization of medical devices (Tables 4 and 5)[69]. The indicated contact times range from 3 hours to 12 hours. However, except for a few of the products, the contact time is based only on the conditions to pass the AOAC Sporicidal Test as a sterilant and not on simulated use testing with devices. These solutions are commonly used as high-level disinfectants when a shorter processing time is required. Generally, chemical liquid sterilants cannot be monitored using a biological indicator to verify sterility[899, 900].

The survival kinetics for thermal sterilization methods, such as steam and dry heat, have been studied and characterized extensively, whereas the kinetics for sterilization with liquid sterilants are less well understood[921]. The information that is available in the literature suggests that sterilization processes based on liquid chemical sterilants, in general, may not convey the same sterility assurance level as sterilization achieved using thermal or physical methods[823]. The data indicate that the survival curves for liquid chemical sterilants may not exhibit log-linear kinetics and the shape of the survivor curve may vary depending of the formulation, chemical nature and stability of the liquid chemical sterilant. In addition, the design of the AOAC Sporicidal Test does not provide quantification of the microbial challenge. Therefore, sterilization with a liquid chemical sterilant may not convey the same sterility assurance as other sterilization methods.

One of the differences between thermal and liquid chemical processes for sterilization of devices is the accessibility of microorganisms to the sterilant. Heat can penetrate barriers, such as biofilm, tissue, and blood, to attain organism kill, whereas liquids cannot adequately penetrate these barriers. In addition, the viscosity of some liquid chemical sterilants impedes their access to organisms in the narrow lumens and mated surfaces of devices[922]. Another limitation to sterilization of devices with liquid chemical germicides is the post-processing environment of the device. Devices cannot be wrapped or adequately contained during processing in a liquid chemical sterilant to maintain sterility following processing and during storage. Furthermore, devices may require rinsing following exposure to the liquid chemical sterilant with water that typically is not sterile. Therefore, due to the inherent limitations of using liquid chemical sterilants, their use should be restricted to reprocessing critical devices that are heat-sensitive and incompatible with other sterilization methods.

Several published studies compare the sporicidal effect of liquid chemical germicides against spores of *Bacillus* and *Clostridium*[78, 659, 660, 715].

Performic Acid. Performic acid is a fast-acting sporicide that was incorporated into an automated endoscope reprocessing system[400]. Systems using performic acid are not currently FDA cleared.

Filtration. Although filtration is not a lethality-based process and is not an FDA-cleared sterilization method, this technology is used to remove bacteria from thermolabile pharmaceutical fluids

that cannot be purified by any other means. In order to remove bacteria, the membrane pore size (e.g., 0.22 μm) must be smaller than the bacteria and uniform throughout[923]. Some investigators have appropriately questioned whether the removal of microorganisms by filtration really is a sterilization method because of slight bacterial passage through filters, viral passage through filters, and transference of the sterile filtrate into the final container under aseptic conditions entail a risk of contamination[924].

Microwave. Microwaves are used in medicine for disinfection of soft contact lenses, dental instruments, dentures, milk, and urinary catheters for intermittent self-catheterization[925-931]. However, microwaves must only be used with products that are compatible (e.g., do not melt) [931]. Microwaves are radio-frequency waves, which are usually used at a frequency of 2450 MHz. The microwaves produce friction of water molecules in an alternating electrical field. The intermolecular friction derived from the vibrations generates heat and some authors believe that the effect of microwaves depends on the heat produced while others postulate a nonthermal lethal effect[932-934]. The initial reports showed microwaves to be an effective microbicide. The microwaves produced by a "home-type" microwave oven (2.45 GHz) completely inactivate bacterial cultures, mycobacteria, viruses, and *G. stearothermophilus* spores within 60 seconds to 5 minutes depending on the challenge organism[933, 935-937]. Another study confirmed these results but also found that higher power microwaves in the presence of water may be needed for sterilization[932]. Complete destruction of *Mycobacterium bovis* was obtained with 4 minutes of microwave exposure (600W, 2450 MHz)[937]. The effectiveness of microwave ovens for different sterilization and disinfection purposes should be tested and demonstrated as test conditions affect the results (e.g., presence of water, microwave power). Sterilization of metal instruments can be accomplished but requires certain precautions.[926]. Of concern is that home-type microwave ovens may not have even distribution of microwave energy over the entire dry device (there may be hot and cold spots on solid medical devices); hence there may be areas that are not sterilized or disinfected. The use of microwave ovens to disinfect intermittent-use catheters also has been suggested. Researchers found that test bacteria (e.g., *E. coli*, *Klebsiella pneumoniae*, *Candida albicans*) were eliminated from red rubber catheters within 5 minutes [931]. Microwaves used for sterilization of medical devices have not been FDA cleared.

Glass Bead "Sterilizer". Glass bead "sterilization" uses small glass beads (1.2-1.5 mm diameter) and high temperature (217 ^{o}C -232^{o}C) for brief exposure times (e.g., 45 seconds) to inactivate microorganisms. These devices have been used for several years in the dental profession[938-940]. FDA believes there is a risk of infection with this device because of potential failure to sterilize dental instruments and their use should be discontinued until the device has received FDA clearance.

Vaporized Hydrogen Peroxide **(VHP®).** Hydrogen peroxide solutions have been used as chemical sterilants for many years. However, the VHP® was not developed for the sterilization of medical equipment until the mid-1980s. One method for delivering VHP to the reaction site uses a deep vacuum to pull liquid hydrogen peroxide (30-35% concentration) from a disposable cartridge through a heated vaporizer and then, following vaporization, into the sterilization chamber. A second approach to VHP delivery is the flow-through approach in which the VHP is carried into the sterilization chamber by a carrier gas such as air using either a slight negative pressure (vacuum) or slight positive pressure. Applications of this technology include vacuum systems for industrial sterilization of medical devices and atmospheric systems for decontaminating for large and small areas[853]. VHP offers several appealing features that include rapid cycle time (e.g., 30-45 minutes); low temperature; environmentally safe by-products (H_2O, oxygen [O_2]); good material compatibility; and ease of operation, installation and monitoring. VHP has limitations including that cellulose cannot be processed; nylon becomes brittle; and VHP penetration capabilities are less than those of ETO. VHP has not been cleared by FDA for sterilization of medical devices in healthcare facilities.

The feasibility of utilizing vapor-phase hydrogen peroxide as a surface decontaminant and sterilizer was evaluated in a centrifuge decontamination application. In this study, vapor-phase hydrogen peroxide was shown to possess significant sporicidal activity [941]. In preliminary studies, hydrogen

peroxide vapor decontamination has been found to be a highly effective method of eradicating MRSA, *Serratia marcescens, Clostridium botulinum spores* and *Clostridium difficile* from rooms, furniture, surfaces and/or equipment; however, further investigation of this method to demonstrate both safety and effectiveness in reducing infection rates are required[942-945].

***Ozone*.** Ozone has been used for years as a drinking water disinfectant. Ozone is produced when O_2 is energized and split into two monatomic (O_1) molecules. The monatomic oxygen molecules then collide with O_2 molecules to form ozone, which is O_3. Thus, ozone consists of O_2 with a loosely bonded third oxygen atom that is readily available to attach to, and oxidize, other molecules. This additional oxygen atom makes ozone a powerful oxidant that destroys microorganisms but is highly unstable (i.e., half-life of 22 minutes at room temperature).

A new sterilization process, which uses ozone as the sterilant, was cleared by FDA in August 2003 for processing reusable medical devices. The sterilizer creates its own sterilant internally from USP grade oxygen, steam-quality water and electricity; the sterilant is converted back to oxygen and water vapor at the end of the cycle by a passing through a catalyst before being exhausted into the room. The duration of the sterilization cycle is about 4 h and 15 m, and it occurs at 30-35^{o}C. Microbial efficacy has been demonstrated by achieving a SAL of 10^{-6} with a variety of microorganisms to include the most resistant microorganism, *Geobacillus stearothermophilus*.

The ozone process is compatible with a wide range of commonly used materials including stainless steel, titanium, anodized aluminum, ceramic, glass, silica, PVC, Teflon, silicone, polypropylene, polyethylene and acrylic. In addition, rigid lumen devices of the following diameter and length can be processed: internal diameter (ID): > 2 mm, length ≤ 25 cm; ID > 3 mm, length ≤ 47 cm; and ID > 4 mm, length ≤ 60 cm.

The process should be safe for use by the operator because there is no handling of the sterilant, no toxic emissions, no residue to aerate, and low operating temperature means there is no danger of an accidental burn. The cycle is monitored using a self-contained biological indicator and a chemical indicator. The sterilization chamber is small, about 4 ft^3 (Written communication, S Dufresne, July 2004).

A gaseous ozone generator was investigated for decontamination of rooms used to house patients colonized with MRSA. The results demonstrated that the device tested would be inadequate for the decontamination of a hospital room[946].

***Formaldehyde Steam*.** Low-temperature steam with formaldehyde is used as a low-temperature sterilization method in many countries, particularly in Scandinavia, Germany, and the United Kingdom. The process involves the use of formalin, which is vaporized into a formaldehyde gas that is admitted into the sterilization chamber. A formaldehyde concentration of 8-16 mg/l is generated at an operating temperature of 70-75^{o}C. The sterilization cycle consists of a series of stages that include an initial vacuum to remove air from the chamber and load, followed by steam admission to the chamber with the vacuum pump running to purge the chamber of air and to heat the load, followed by a series of pulses of formaldehyde gas, followed by steam. Formaldehyde is removed from the sterilizer and load by repeated alternate evacuations and flushing with steam and air. This system has some advantages, e.g., the cycle time for formaldehyde gas is faster than that for ETO and the cost per cycle is relatively low. However, ETO is more penetrating and operates at lower temperatures than do steam/formaldehyde sterilizers. Low-temperature steam formaldehyde sterilization has been found effective against vegetative bacteria, mycobacteria, *B. atrophaeus* and *G. stearothermophilus* spores and *Candida albicans*[947-949].

Formaldehyde vapor cabinets also may be used in healthcare facilities to sterilize heat-sensitive medical equipment[950]. Commonly, there is no circulation of formaldehyde and no temperature and humidity controls. The release of gas from paraformaldehyde tablets (placed on the lower tray) is slow and produces a low partial pressure of gas. The microbicidal quality of this procedure is unknown[951].

Reliable sterilization using formaldehyde is achieved when performed with a high concentration of gas, at a temperature between 60° and 80°C and with a relative humidity of 75 to 100%.

Studies indicate that formaldehyde is a mutagen and a potential human carcinogen, and OSHA regulates formaldehyde. The permissible exposure limit for formaldehyde in work areas is 0.75 ppm measured as a 8-hour TWA. The OSHA standard includes a 2 ppm STEL (i.e., maximum exposure allowed during a 15-minute period). As with the ETO standard, the formaldehyde standard requires that the employer conduct initial monitoring to identify employees who are exposed to formaldehyde at or above the action level or STEL. If this exposure level is maintained, employers may discontinue exposure monitoring until there is a change that could affect exposure levels or an employee reports formaldehyde-related signs and symptoms[269, 578]. The formaldehyde steam sterilization system has not been FDA cleared for use in healthcare facilities.

Gaseous chlorine dioxide. A gaseous chlorine dioxide system for sterilization of healthcare products was developed in the late 1980s[853, 952, 953]. Chlorine dioxide is not mutagenic or carcinogenic in humans. As the chlorine dioxide concentration increases, the time required to achieve sterilization becomes progressively shorter. For example, only 30 minutes were required at 40 mg/l to sterilize the 10^6 *B. atrophaeus* spores at 30° to 32°C[954]. Currently, no gaseous chlorine dioxide system is FDA cleared.

Vaporized Peracetic Acid. The sporicidal activity of peracetic acid vapor at 20, 40, 60, and 80% relative humidity and 25°C was determined on *Bacillus atrophaeus* spores on paper and glass surfaces. Appreciable activity occurred within 10 minutes of exposure to 1 mg of peracetic acid per liter at 40% or higher relative humidity[955]. No vaporized peracetic acid system is FDA cleared.

Infrared radiation. An infrared radiation prototype sterilizer was investigated and found to destroy *B. atrophaeus* spores. Some of the possible advantages of infrared technology include short cycle time, low energy consumption, no cycle residuals, and no toxicologic or environmental effects. This may provide an alternative technology for sterilization of selected heat-resistant instruments but there are no FDA-cleared systems for use in healthcare facilities [956].

The other sterilization technologies mentioned above may be used for sterilization of critical medical items if cleared by the FDA and ideally, the microbicidal effectiveness of the technology has been published in the scientific literature. The selection and use of disinfectants, chemical sterilants and sterilization processes in the healthcare field is dynamic, and products may become available that are not in existence when this guideline was written. As newer disinfectants and sterilization processes become available, persons or committees responsible for selecting disinfectants and sterilization processes should be guided by products cleared by FDA and EPA as well as information in the scientific literature.

Sterilizing Practices

Overview. The delivery of sterile products for use in patient care depends not only on the effectiveness of the sterilization process but also on the unit design, decontamination, disassembling and packaging of the device, loading the sterilizer, monitoring, sterilant quality and quantity, and the appropriateness of the cycle for the load contents, and other aspects of device reprocessing. Healthcare personnel should perform most cleaning, disinfecting, and sterilizing of patient-care supplies in a central processing department in order to more easily control quality. The aim of central processing is the orderly processing of medical and surgical instruments to protect patients from infections while minimizing risks to staff and preserving the value of the items being reprocessed[957]. Healthcare facilities should promote the same level of efficiency and safety in the preparation of supplies in other areas (e.g., operating room, respiratory therapy) as is practiced in central processing.

Ensuring consistency of sterilization practices requires a comprehensive program that ensures operator competence and proper methods of cleaning and wrapping instruments, loading the sterilizer,

operating the sterilizer, and monitoring of the entire process. Furthermore, care must be consistent from an infection prevention standpoint in all patient-care settings, such as hospital and outpatient facilities.

Sterilization Cycle Verification. A sterilization process should be verified before it is put into use in healthcare settings. All steam, ETO, and other low-temperature sterilizers are tested with biological and chemical indicators upon installation, when the sterilizer is relocated, redesigned, after major repair and after a sterilization failure has occurred to ensure they are functioning prior to placing them into routine use. Three consecutive empty steam cycles are run with a biological and chemical indicator in an appropriate test package or tray. Each type of steam cycle used for sterilization (e.g., vacuum-assisted, gravity) is tested separately. In a prevacuum steam sterilizer three consecutive empty cycles are also run with a Bowie-Dick test. The sterilizer is not put back into use until all biological indicators are negative and chemical indicators show a correct end-point response[811-814, 819, 958].

Biological and chemical indicator testing is also done for ongoing quality assurance testing of representative samples of actual products being sterilized and product testing when major changes are made in packaging, wraps, or load configuration. Biological and chemical indicators are placed in products, which are processed in a full load. When three consecutive cycles show negative biological indicators and chemical indicators with a correct end point response, you can put the change made into routine use[811-814, 958]. Items processed during the three evaluation cycles should be quarantined until the test results are negative.

Physical Facilities. The central processing area(s) ideally should be divided into at least three areas: decontamination, packaging, and sterilization and storage. Physical barriers should separate the decontamination area from the other sections to contain contamination on used items. In the decontamination area reusable contaminated supplies (and possibly disposable items that are reused) are received, sorted, and decontaminated. The recommended airflow pattern should contain contaminates within the decontamination area and minimize the flow of contaminates to the clean areas. The American Institute of Architects [959] recommends negative pressure and no fewer than six air exchanges per hour in the decontamination area (AAMI recommends 10 air changes per hour) and 10 air changes per hour with positive pressure in the sterilizer equipment room. The packaging area is for inspecting, assembling, and packaging clean, but not sterile, material. The sterile storage area should be a limited access area with a controlled temperature (may be as high as 75°F) and relative humidity (30-60% in all works areas except sterile storage, where the relative humidity should not exceed 70%)[819]. The floors and walls should be constructed of materials capable of withstanding chemical agents used for cleaning or disinfecting. Ceilings and wall surfaces should be constructed of non-shedding materials. Physical arrangements of processing areas are presented schematically in four references[811, 819, 920, 957].

Cleaning. As repeatedly mentioned, items must be cleaned using water with detergents or enzymatic cleaners [465, 466, 468] before processing. Cleaning reduces the bioburden and removes foreign material (i.e., organic residue and inorganic salts) that interferes with the sterilization process by acting as a barrier to the sterilization agent[179, 426, 457, 911, 912]. Surgical instruments are generally presoaked or prerinsed to prevent drying of blood and tissue. Precleaning in patient-care areas may be needed on items that are heavily soiled with feces, sputum, blood, or other material. Items sent to central processing without removing gross soil may be difficult to clean because of dried secretions and excretions. Cleaning and decontamination should be done as soon as possible after items have been used.

Several types of mechanical cleaning machines (e.g., utensil washer-sanitizer, ultrasonic cleaner, washer-sterilizer, dishwasher, washer-disinfector) may facilitate cleaning and decontamination of most items. This equipment often is automated and may increase productivity, improve cleaning effectiveness, and decrease worker exposure to blood and body fluids. Delicate and intricate objects and heat- or moisture-sensitive articles may require careful cleaning by hand. All used items sent to the central processing area should be considered contaminated (unless decontaminated in the area of origin), handled with gloves (forceps or tongs are sometimes needed to avoid exposure to sharps), and decontaminated by one of the aforementioned methods to render them safer to handle. Items composed

of more than one removable part should be disassembled. Care should be taken to ensure that all parts are kept together, so that reassembly can be accomplished efficiently[811].

Investigators have described the degree of cleanliness by visual and microscopic examination. One study found 91% of the instruments to be clean visually but, when examined microscopically, 84% of the instruments had residual debris. Sites that contained residual debris included junctions between insulating sheaths and activating mechanisms of laparoscopic instruments and articulations and grooves of forceps. More research is needed to understand the clinical significance of these findings [960] and how to ensure proper cleaning.

Personnel working in the decontamination area should wear household-cleaning-type rubber or plastic gloves when handling or cleaning contaminated instruments and devices. Face masks, eye protection such as goggles or full-length faceshields, and appropriate gowns should be worn when exposure to blood and contaminated fluids may occur (e.g., when manually cleaning contaminated devices)[961]. Contaminated instruments are a source of microorganisms that could inoculate personnel through nonintact skin on the hands or through contact with the mucous membranes of eyes, nose, or mouth[214, 811, 813]. Reusable sharps that have been in contact with blood present a special hazard. Employees must not reach with their gloved hands into trays or containers that hold these sharps to retrieve them[214]. Rather, employees should use engineering controls (e.g., forceps) to retrieve these devices.

Packaging. Once items are cleaned, dried, and inspected, those requiring sterilization must be wrapped or placed in rigid containers and should be arranged in instrument trays/baskets according to the guidelines provided by the AAMI and other professional organizations[454, 811-814, 819, 836, 962]. These guidelines state that hinged instruments should be opened; items with removable parts should be disassembled unless the device manufacturer or researchers provide specific instructions or test data to the contrary[181]; complex instruments should be prepared and sterilized according to device manufacturer's instructions and test data; devices with concave surfaces should be positioned to facilitate drainage of water; heavy items should be positioned not to damage delicate items; and the weight of the instrument set should be based on the design and density of the instruments and the distribution of metal mass[811, 962]. While there is no longer a specified sterilization weight limit for surgical sets, heavy metal mass is a cause of wet packs (i.e., moisture inside the case and tray after completion of the sterilization cycle)[963]. Other parameters that may influence drying are the density of the wraps and the design of the set[964].

There are several choices in methods to maintain sterility of surgical instruments, including rigid containers, peel-open pouches (e.g., self-sealed or heat-sealed plastic and paper pouches), roll stock or reels (i.e., paper-plastic combinations of tubing designed to allow the user to cut and seal the ends to form a pouch) [454] and sterilization wraps (woven and nonwoven). Healthcare facilities may use all of these packaging options. The packaging material must allow penetration of the sterilant, provide protection against contact contamination during handling, provide an effective barrier to microbial penetration, and maintain the sterility of the processed item after sterilization [965]. An ideal sterilization wrap would successfully address barrier effectiveness, penetrability (i.e., allows sterilant to penetrate), aeration (e.g., allows ETO to dissipate), ease of use, drapeability, flexibility, puncture resistance, tear strength, toxicity, odor, waste disposal, linting, cost, and transparency[966]. Unacceptable packaging for use with ETO (e.g., foil, polyvinylchloride, and polyvinylidene chlorine [kitchen-type transparent wrap]) [814] or hydrogen peroxide gas plasma (e.g., linens and paper) should not be used to wrap medical items.

In central processing, double wrapping can be done sequentially or nonsequentially (i.e., simultaneous wrapping). Wrapping should be done in such a manner to avoid tenting and gapping. The sequential wrap uses two sheets of the standard sterilization wrap, one wrapped after the other. This procedure creates a package within a package. The nonsequential process uses two sheets wrapped at the same time so that the wrapping needs to be performed only once. This latter method provides

multiple layers of protection of surgical instruments from contamination and saves time since wrapping is done only once. Multiple layers are still common practice due to the rigors of handling within the facility even though the barrier efficacy of a single sheet of wrap has improved over the years[966]. Written and illustrated procedures for preparation of items to be packaged should be readily available and used by personnel when packaging procedures are performed[454].

***Loading*.** All items to be sterilized should be arranged so all surfaces will be directly exposed to the sterilizing agent. Thus, loading procedures must allow for free circulation of steam (or another sterilant) around each item. Historically, it was recommended that muslin fabric packs should not exceed the maximal dimensions, weight, and density of 12 inches wide x 12 inches high x 20 inches long, 12 lbs, and 7.2 lbs per cubic foot, respectively. Due to the variety of textiles and metal/plastic containers on the market, the textile and metal/plastic container manufacturer and the sterilizer manufacturers should be consulted for instructions on pack preparation and density parameters[819].

There are several important basic principles for loading a sterilizer: allow for proper sterilant circulation; perforated trays should be placed so the tray is parallel to the shelf; nonperforated containers should be placed on their edge (e.g., basins); small items should be loosely placed in wire baskets; and peel packs should be placed on edge in perforated or mesh bottom racks or baskets[454, 811, 836].

***Storage*.** Studies in the early 1970s suggested that wrapped surgical trays remained sterile for varying periods depending on the type of material used to wrap the trays. Safe storage times for sterile packs vary with the porosity of the wrapper and storage conditions (e.g., open versus closed cabinets). Heat-sealed, plastic peel-down pouches and wrapped packs sealed in 3-mil (3/1000 inch) polyethylene overwrap have been reported to be sterile for as long as 9 months after sterilization. The 3-mil polyethylene is applied after sterilization to extend the shelf life for infrequently used items[967]. Supplies wrapped in double-thickness muslin comprising four layers, or equivalent, remain sterile for at least 30 days. Any item that has been sterilized should not be used after the expiration date has been exceeded or if the sterilized package is wet, torn, or punctured.

Although some hospitals continue to date every sterilized product and use the time-related shelf-life practice, many hospitals have switched to an event-related shelf-life practice. This latter practice recognizes that the product should remain sterile until some event causes the item to become contaminated (e.g., tear in packaging, packaging becomes wet, seal is broken)[968]. Event-related factors that contribute to the contamination of a product include bioburden (i.e., the amount of contamination in the environment), air movement, traffic, location, humidity, insects, vermin, flooding, storage area space, open/closed shelving, temperature, and the properties of the wrap material[966, 969]. There are data that support the event-related shelf-life practice[970-972]. One study examined the effect of time on the sterile integrity of paper envelopes, peel pouches, and nylon sleeves. The most important finding was the absence of a trend toward an increased rate of contamination over time for any pack when placed in covered storage[971]. Another evaluated the effectiveness of event-related outdating by microbiologically testing sterilized items. During the 2-year study period, all of the items tested were sterile[972]. Thus, contamination of a sterile item is event-related and the probability of contamination increases with increased handling[973].

Following the sterilization process, medical and surgical devices must be handled using aseptic technique in order to prevent contamination. Sterile supplies should be stored far enough from the floor (8 to 10 inches), the ceiling (5 inches unless near a sprinkler head [18 inches from sprinkler head]), and the outside walls (2 inches) to allow for adequate air circulation, ease of cleaning, and compliance with local fire codes (e.g., supplies must be at least 18 inches from sprinkler heads). Medical and surgical supplies should not be stored under sinks or in other locations where they can become wet. Sterile items that become wet are considered contaminated because moisture brings with it microorganisms from the air and surfaces. Closed or covered cabinets are ideal but open shelving may be used for storage. Any package that has fallen or been dropped on the floor must be inspected for damage to the packaging and

contents (if the items are breakable). If the package is heat-sealed in impervious plastic and the seal is still intact, the package should be considered not contaminated. If undamaged, items packaged in plastic need not be reprocessed.

Monitoring. The sterilization procedure should be monitored routinely by using a combination of mechanical, chemical, and biological indicators to evaluate the sterilizing conditions and indirectly the microbiologic status of the processed items. The mechanical monitors for steam sterilization include the daily assessment of cycle time and temperature by examining the temperature record chart (or computer printout) and an assessment of pressure via the pressure gauge. The mechanical monitors for ETO include time, temperature, and pressure recorders that provide data via computer printouts, gauges, and/or displays[814]. Generally, two essential elements for ETO sterilization (i.e., the gas concentration and humidity) cannot be monitored in healthcare ETO sterilizers.

Chemical indicators are convenient, are inexpensive, and indicate that the item has been exposed to the sterilization process. In one study, chemical indicators were more likely than biological indicators to inaccurately indicate sterilization at marginal sterilization times (e.g., 2 minutes)[847]. Chemical indicators should be used in conjunction with biological indicators, but based on current studies should not replace them because they indicate sterilization at marginal sterilization time and because only a biological indicator consisting of resistant spores can measure the microbial killing power of the sterilization process.[847, 974]. Chemical indicators are affixed on the outside of each pack to show that the package has been processed through a sterilization cycle, but these indicators do not prove sterilization has been achieved. Preferably, a chemical indicator also should be placed on the inside of each pack to verify sterilant penetration. Chemical indicators usually are either heat-or chemical-sensitive inks that change color when one or more sterilization parameters (e.g., steam-time, temperature, and/or saturated steam; ETO-time, temperature, relative humidity and/or ETO concentration) are present. Chemical indicators have been grouped into five classes based on their ability to monitor one or multiple sterilization parameters[813, 819]. If the internal and/or external indicator suggests inadequate processing, the item should not be used[815]. An air-removal test (Bowie-Dick Test) must be performed daily in an empty dynamic-air-removal sterilizer (e.g., prevacuum steam sterilizer) to ensure air removal.

Biological indicators are recognized by most authorities as being closest to the ideal monitors of the sterilization process [974, 975] because they measure the sterilization process directly by using the most resistant microorganisms (i.e., *Bacillus* spores), and not by merely testing the physical and chemical conditions necessary for sterilization. Since the *Bacillus* spores used in biological indicators are more resistant and present in greater numbers than are the common microbial contaminants found on patient-care equipment, the demonstration that the biological indicator has been inactivated strongly implies that other potential pathogens in the load have been killed[844].

An ideal biological monitor of the sterilization process should be easy to use, be inexpensive, not be subject to exogenous contamination, provide positive results as soon as possible after the cycle so that corrective action may be accomplished, and provide positive results only when the sterilization parameters (e.g., steam-time, temperature, and/or saturated steam; ETO-time, temperature, relative humidity and/or ETO concentration) are inadequate to kill microbial contaminates[847].

Biological indicators are the only process indicators that directly monitor the lethality of a given sterilization process. Spores used to monitor a sterilization process have demonstrated resistance to the sterilizing agent and are more resistant than the bioburden found on medical devices[179, 911, 912]. *B. atrophaeus* spores (10^6) are used to monitor ETO and dry heat, and *G. stearothermophilus* spores (10^5) are used to monitor steam sterilization, hydrogen peroxide gas plasma, and liquid peracetic acid sterilizers. *G. stearothermophilus* is incubated at 55-60°C, and *B. atrophaeus* is incubated at 35-37°C. Steam and low temperature sterilizers (e.g., hydrogen peroxide gas plasma, peracetic acid) should be monitored at least weekly with the appropriate commercial preparation of spores. If a sterilizer is used frequently (e.g., several loads per day), daily use of biological indicators allows earlier discovery of

equipment malfunctions or procedural errors and thus minimizes the extent of patient surveillance and product recall needed in the event of a positive biological indicator[811]. Each load should be monitored if it contains implantable objects. If feasible, implantable items should not be used until the results of spore tests are known to be negative.

Originally, spore-strip biological indicators required up to 7 days of incubation to detect viable spores from marginal cycles (i.e., when few spores remained viable). The next generation of biological indicator was self-contained in plastic vials containing a spore-coated paper strip and a growth media in a crushable glass ampoule. This indicator had a maximum incubation of 48 hours but significant failures could be detected in ≤24 hours. A rapid-readout biological indicator that detects the presence of enzymes of *G. stearothermophilus* by reading a fluorescent product produced by the enzymatic breakdown of a nonfluorescent substrate has been marketed for the more than 10 years. Studies demonstrate that the sensitivity of rapid-readout tests for steam sterilization (1 hour for 132°C gravity sterilizers, 3 hrs for 121°C gravity and 132°C vacuum sterilizers) parallels that of the conventional sterilization-specific biological indicators [846, 847, 976, 977] and the fluorescent rapid readout results reliably predict 24- and 48-hour and 7-day growth[978]. The rapid-readout biological indicator is a dual indicator system as it also detects acid metabolites produced during growth of the *G. stearothermophilus* spores. This system is different from the indicator system consisting of an enzyme system of bacterial origin without spores. Independent comparative data using suboptimal sterilization cycles (e.g., reduced time or temperature) with the enzyme-based indicator system have not been published[979].

A new rapid-readout ETO biological indicator has been designed for rapid and reliable monitoring of ETO sterilization processes. The indicator has been cleared by the FDA for use in the United States[400]. The rapid-readout ETO biological indicator detects the presence of *B. atrophaeus* by detecting a fluorescent signal indicating the activity of an enzyme present within the *B. atrophaeus* organism, beta-glucosidase. The fluorescence indicates the presence of an active spore-associated enzyme and a sterilization process failure. This indicator also detects acid metabolites produced during growth of the *B. atrophaeus* spore. Per manufacturer's data, the enzyme always was detected whenever viable spores were present. This was expected because the enzyme is relatively ETO resistant and is inactivated at a slightly longer exposure time than the spore. The rapid-readout ETO biological indicator can be used to monitor 100% ETO, and ETO-HCFC mixture sterilization cycles. It has not been tested in ETO-CO_2 mixture sterilization cycles.

The standard biological indicator used for monitoring full-cycle steam sterilizers does not provide reliable monitoring flash sterilizers[980]. Biological indicators specifically designed for monitoring flash sterilization are now available, and studies comparing them have been published[846, 847, 981].

Since sterilization failure can occur (about 1% for steam)[982], a procedure to follow in the event of positive spore tests with steam sterilization has been provided by CDC and the Association of periOperative Registered Nurses (AORN). The 1981 CDC recommendation is that "objects, other than implantable objects, do not need to be recalled because of a single positive spore test unless the steam sterilizer or the sterilization procedure is defective." The rationale for this recommendation is that single positive spore tests in sterilizers occur sporadically. They may occur for reasons such as slight variation in the resistance of the spores[983], improper use of the sterilizer, and laboratory contamination during culture (uncommon with self-contained spore tests). If the mechanical (e.g., time, temperature, pressure in the steam sterilizer) and chemical (internal and/or external) indicators suggest that the sterilizer was functioning properly, a single positive spore test probably does not indicate sterilizer malfunction but the spore test should be repeated immediately [983]. If the spore tests remain positive, use of the sterilizer should be discontinued until it is serviced[1]. Similarly, AORN states that a single positive spore test does not necessarily indicate a sterilizer failure. If the test is positive, the sterilizer should immediately be rechallenged for proper use and function. Items, other than implantable ones, do not necessarily need to be recalled unless a sterilizer malfunction is found. If a sterilizer malfunction is discovered, the items must be considered nonsterile, and the items from the suspect load(s) should be recalled, insofar as

possible, and reprocessed [984]. A suggested protocol for management of positive biological indicators is shown in Table 12[839]. A more conservative approach also has been recommended [813] in which any positive spore test is assumed to represent sterilizer malfunction and requires that all materials processed in that sterilizer, dating from the sterilization cycle having the last negative biologic indicator to the next cycle showing satisfactory biologic indicator challenge results, must be considered nonsterile and retrieved, if possible, and reprocessed. This more conservative approach should be used for sterilization methods other than steam (e.g., ETO, hydrogen peroxide gas plasma). However, no action is necessary if there is strong evidence for the biological indicator being defective [983] or the growth medium contained a *Bacillus* contaminant[985] .

If patient-care items were used before retrieval, the infection control professional should assess the risk of infection in collaboration with central processing, surgical services, and risk management staff. The factors that should be considered include the chemical indicator result (e.g., nonreactive chemical indicator may indicate temperature not achieved); the results of other biological indicators that followed the positive biological indicator (e.g., positive on Tuesday, negative on Wednesday); the parameters of the sterilizer associated with the positive biological indicator (e.g., reduced time at correct temperature); the time-temperature chart (or printout); and the microbial load associated with decontaminated surgical instruments (e.g., 85% of decontaminated surgical instruments have less than 100 CFU). The margin of safety in steam sterilization is sufficiently large that there is minimal infection risk associated with items in a load that show spore growth, especially if the item was properly cleaned and the temperature was achieved (e.g., as shown by acceptable chemical indicator or temperature chart). There are no published studies that document disease transmission via a nonretrieved surgical instrument following a sterilization cycle with a positive biological indicator.

False-positive biological indicators may occur from improper testing or faulty indicators. The latter may occur from improper storage, processing, product contamination, material failure, or variation in resistance of spores. Gram stain and subculture of a positive biological indicator may determine if a contaminant has created a false-positive result[839, 986]. However, in one incident, the broth used as growth medium contained a contaminant, *B. coagulans,* which resulted in broth turbidity at 55°C[985]. Testing of paired biological indicators from different manufacturers can assist in assessing a product defect[839]. False-positive biological indicators due to extrinsic contamination when using self-contained biological indicators should be uncommon. A biological indicator should not be considered a false-positive indicator until a thorough analysis of the entire sterilization process shows this to be likely.

The size and composition of the biological indicator test pack should be standardized to create a significant challenge to air removal and sterilant penetration and to obtain interpretable results. There is a standard 16-towel pack recommended by AAMI for steam sterilization [813, 819, 987] consisting of 16 clean, preconditioned, reusable huck or absorbent surgical towels each of which is approximately 16 inches by 26 inches. Each towel is folded lengthwise into thirds and then folded widthwise in the middle. One or more biological indicators are placed between the eight and ninth towels in the approximate geometric center of the pack. When the towels are folded and placed one on top of another, to form a stack (approximately 6 inch height) it should weigh approximately 3 pounds and should have a density of approximately 11.3 pounds per cubic foot[813]. This test pack has not gained universal use as a standard pack that simulates the actual in-use conditions of steam sterilizers. Commercially available disposable test packs that have been shown to be equivalent to the AAMI 16 towel test pack also may be used. The test pack should be placed flat in an otherwise fully loaded sterilizer chamber, in the area least favorable to sterilization (i.e., the area representing the greatest challenge to the biological indicator). This area is normally in the front, bottom section of the sterilizer, near the drain[811, 813]. A control biological indicator from the lot used for testing should be left unexposed to the sterilant, and then incubated to verify the presterilization viability of the test spores and proper incubation. The most conservative approach would be to use a control for each run; however, less frequent use may be adequate (e.g., weekly). There also is a routine test pack for ETO where a biological indicator is placed in a plastic syringe with plunger, then placed in the folds of a clean surgical towel, and wrapped. Alternatively, commercially available disposal

test packs that have been shown to be equivalent to the AAMI test pack may be used. The test pack is placed in the center of the sterilizer load[814]. Sterilization records (mechanical, chemical, and biological) should be retained for a time period in compliance with standards (e.g., Joint Commission for the Accreditation of Healthcare Facilities requests 3 years) and state and federal regulations.

In Europe, biological monitors are not used routinely to monitor the sterilization process. Instead, release of sterilizer items is based on monitoring the physical conditions of the sterilization process that is termed "parametric release." Parametric release requires that there is a defined quality system in place at the facility performing the sterilization and that the sterilization process be validated for the items being sterilized. At present in Europe, parametric release is accepted for steam, dry heat, and ionizing radiation processes, as the physical conditions are understood and can be monitored directly[988]. For example, with steam sterilizers the load could be monitored with probes that would yield data on temperature, time, and humidity at representative locations in the chamber and compared to the specifications developed during the validation process.

Periodic infection control rounds to areas using sterilizers to standardize the sterilizer's use may identify correctable variances in operator competence; documentation of sterilization records, including chemical and biological indicator test results; sterilizer maintenance and wrapping; and load numbering of packs. These rounds also may identify improvement activities to ensure that operators are adhering to established standards[989].

REUSE OF SINGLE-USE MEDICAL DEVICES

The reuse of single-use medical devices began in the late 1970s. Before this time most devices were considered reusable. Reuse of single-use devices increased as a cost-saving measure. Approximately 20 to 30% of U.S. hospitals reported that they reuse at least one type of single-use device. Reuse of single-use devices involves regulatory, ethical, medical, legal and economic issues and has been extremely controversial for more than two decades[990]. The U.S. public has expressed increasing concern regarding the risk of infection and injury when reusing medical devices intended and labeled for single use. Although some investigators have demonstrated it is safe to reuse disposable medical devices such as cardiac electrode catheters, [991-993] additional studies are needed to define the risks [994] and document the benefits. In August 2000, FDA released a guidance document on single-use devices reprocessed by third parties or hospitals[995]. In this guidance document, FDA states that hospitals or third-party reprocessors will be considered "manufacturers" and regulated in the same manner. A reused single-use device will have to comply with the same regulatory requirements of the device when it was originally manufactured. This document presents FDA's intent to enforce premarket submission requirements within 6 months (February 2001) for class III devices (e.g., cardiovascular intra-aortic balloon pump, transluminal coronary angioplasty catheter); 12 months (August 2001) for class II devices (e.g., blood pressure cuff, bronchoscope biopsy forceps); and 18 months (February 2002) for class I devices (e.g., disposable medical scissors, ophthalmic knife). FDA uses two types of premarket requirements for nonexempt class I and II devices, a 510(k) submission that may have to show that the device is as safe and effective as the same device when new, and a premarket approval application. The 510(k) submission must provide scientific evidence that the device is safe and effective for its intended use. FDA allowed hospitals a year to comply with the nonpremarket requirements (registration and listing, reporting adverse events associated with medical devices, quality system regulations, and proper labeling). The options for hospitals are to stop reprocessing single-use devices, comply with the rule, or outsource to a third-party reprocessor. FDA guidance document does not apply to permanently implantable pacemakers, hemodialyzers, opened but unused single-use devices, or healthcare settings other than acute-care hospitals. The reuse of single use medical devices continues to be an evolving area of regulations. For this reason, healthcare workers should refer to FDA for the latest guidance (www.fda.gov)[996].

CONCLUSION

When properly used, disinfection and sterilization can ensure the safe use of invasive and non-invasive medical devices. However, current disinfection and sterilization guidelines must be strictly followed.

WED-BASED DISINFECTION AND STERILIZATION RESOURCES

Additional information about disinfection and sterilization is available at the following dedicated websites:

Food and Drug Administration, Rockville, Maryland
http://www.fda.gov/dcrh/ode/germlab.html

Environmental Protection Agency, Washington, D.C.
http://www.epa.gov/oppad001/chemregindex.htm

Centers for Disease Control and Prevention, Atlanta, Georgia
http://www.cdc.gov/ncidod/dhqp/sterile.html

University of North Carolina, Chapel Hill, North Carolina
http://www.disinfectionandsterilization.org

RECOMMENDATIONS FOR DISINFECTION AND STERILIZATION IN HEALTHCARE FACILITIES

A. Rationale

The ultimate goal of the Recommendations for Disinfection and Sterilization in Health-Care Facilities, 2008, is to reduce rates of health-care–associated infections through appropriate use of both disinfection and sterilization. Each recommendation is categorized according to scientific evidence, theoretical rationale, applicability, and federal regulations. Examples are included in some recommendations to aid the reader; however, these examples are not intended to define the only method of implementing the recommendation. The CDC system for categorizing recommendations is defined in the following (Rankings) section.

B. Rankings

Category IA. Strongly recommended for implementation and strongly supported by well-designed experimental, clinical, or epidemiologic studies.

Category IB. Strongly recommended for implementation and supported by some experimental, clinical, or epidemiologic studies, and by a strong theoretical rationale.

Category IC. Required by state or federal regulations. Because of state differences, readers should not assume that the absence of an *IC* recommendation implies the absence of state regulations.

Category II. Suggested for implementation and supported by suggestive clinical or epidemiologic studies or by a theoretical rationale.

No recommendation. Unresolved issue. These include practices for which insufficient evidence or no consensus exists regarding efficacy.

C. Recommendations

1. Occupational Health and Exposure

a. Inform each worker of the possible health effects of his or her exposure to infectious agents (e.g., hepatitis B virus [HBV], hepatitis C virus, human immunodeficiency virus [HIV]), and/or chemicals (e.g., EtO, formaldehyde). The information should be consistent with Occupational Safety and Health Administration (OSHA) requirements and identify the areas and tasks in which potential exists for exposure. *Category II, IC*[214, 320, 959, 997, 998]

b. Educate health-care workers in the selection and proper use of personal protective equipment (PPE). *Category II, IC*

c. Ensure that workers wear appropriate PPE to preclude exposure to infectious agents or chemicals through the respiratory system, skin, or mucous membranes of the eyes, nose, or mouth. PPE can include gloves, gowns, masks, and eye protection. The exact type of PPE depends on the infectious or chemical agent and the anticipated duration of exposure. The employer is responsible for making such equipment and training available. *Category II, IC*. [214, 997-999]

d. Establish a program for monitoring occupational exposure to regulated chemicals (e.g., formaldehyde, EtO) that adheres to state and federal regulations. *Category II, IC*. [997, 1000, 1001]

e. Exclude healthcare workers with weeping dermatitis of hands from direct contact with patient-care equipment. *Category IB*. [1002, 1003]

2. Cleaning of Patient-Care Devices

a. In hospitals, perform most cleaning, disinfection, and sterilization of patient-care devices in a central processing department in order to more easily control quality. *Category II*. [454, 836, 959]

b. Meticulously clean patient-care items with water and detergent, or with water and enzymatic cleaners before high-level disinfection or sterilization procedures. *Category IB*. [6, 83, 101, 104-106, 124, 179, 424-426, 436, 465, 471, 911-913, 1004]

i. Remove visible organic residue (e.g., residue of blood and tissue) and inorganic salts with cleaning. Use cleaning agents that are capable of removing visible organic and inorganic residues. *Category IB*. [424-426, 466, 468, 469, 471, 908, 910]

ii. Clean medical devices as soon as practical after use (e.g., at the point of use) because soiled materials become dried onto the instruments. Dried or baked materials on the instrument make the removal process more difficult and the disinfection or sterilization process less effective or ineffective. *Category IB*. [55, 56, 59, 291, 465, 1005, 1006]

c. Perform either manual cleaning (i.e., using friction) or mechanical cleaning (e.g., with ultrasonic cleaners, washer-disinfector, washer-sterilizers). *Category IB*. [426, 456, 471, 999]

d. If using an automatic washer/disinfector, ensure that the unit is used in accordance with the manufacturer's recommendations. *Category IB*. [7, 133, 155, 725]

e. Ensure that the detergents or enzymatic cleaners selected are compatible with the metals and other materials used in medical instruments. Ensure that the rinse step is adequate for removing cleaning residues to levels that will not interfere with subsequent disinfection/sterilization processes. *Category II*. [836, 1004]

f. Inspect equipment surfaces for breaks in integrity that would impair either cleaning or disinfection/sterilization. Discard or repair equipment that no longer functions as intended or cannot be properly cleaned, and disinfected or sterilized. *Category II*. [888]

g.

3. Indications for Sterilization, High-Level Disinfection, and Low-Level Disinfection

a. Before use on each patient, sterilize critical medical and surgical devices and instruments that enter normally sterile tissue or the vascular system or through which a sterile body fluid flows (e.g., blood). See recommendation 7g for exceptions. *Category IA*. [179, 497, 821, 822, 907, 911, 912]

b. Provide, at a minimum, high-level disinfection for semicritical patient-care equipment (e.g., gastrointestinal endoscopes, endotracheal tubes, anesthesia breathing circuits, and respiratory therapy equipment) that touches either mucous membranes or nonintact skin. *Category IA*. [6-8, 17, 20, 99, 101, 108, 113-115, 129, 138, 139, 147, 152-154, 471, 1007]

c. Perform low-level disinfection for noncritical patient-care surfaces (e.g., bedrails, over-the-bed table) and equipment (e.g., blood pressure cuff) that touch intact skin (see Recommendation 5g). *Category II*. [17, 46-48, 50-52, 67, 68, 372, 373, 378, 382, 401]

4. Selection and Use of Low-Level Disinfectants for Noncritical Patient-Care Devices

a. Process noncritical patient-care devices using a disinfectant and the concentration of germicide listed in Table 1. *Category IB*. [17, 46-48, 50-52, 67, 68, 378, 382, 401]

b. Disinfect noncritical medical devices (e.g., blood pressure cuff) with an EPA-registered hospital disinfectant using the label's safety precautions and use directions. Most EPA-registered hospital disinfectants have a label contact time of 10 minutes. However, multiple scientific studies have demonstrated the efficacy of hospital disinfectants against pathogens with a contact time of at least 1 minute. By law, all applicable label instructions on EPA-registered products must be followed. If the user selects exposure conditions that differ from those on the EPA-registered product label, the user assumes liability from any injuries resulting from off-label use and is potentially subject to enforcement action under FIFRA. *Category IB*. [17, 47, 48, 50, 51, 53-57, 59, 60, 62-64, 355, 378, 382]

c. Ensure that, at a minimum, noncritical patient-care devices are disinfected when visibly soiled and on a regular basis (such as after use on each patient or once daily or once weekly). *Category II*. [378, 380, 1008]

d. If dedicated, disposable devices are not available, disinfect noncritical patient-care equipment after using it on a patient who is on contact precautions before using this equipment on another patient. *Category IB*. [47, 67, 391, 1009]

5. Cleaning and Disinfecting Environmental Surfaces in Healthcare Facilities

a. Clean housekeeping surfaces (e.g., floors, tabletops) on a regular basis, when spills occur, and when these surfaces are visibly soiled. *Category II*. [23, 378, 380, 382, 1008, 1010]

b. Disinfect (or clean) environmental surfaces on a regular basis (e.g., daily, three times per week) and when surfaces are visibly soiled. *Category II*. [378, 380, 402, 1008]

c. Follow manufacturers' instructions for proper use of disinfecting (or detergent) products --- such as recommended use-dilution, material compatibility, storage, shelf-life, and safe use and

disposal. *Category II*. [327, 365, 404]

d. Clean walls, blinds, and window curtains in patient-care areas when these surfaces are visibly contaminated or soiled. *Category II*. [1011]
e. Prepare disinfecting (or detergent) solutions as needed and replace these with fresh solution frequently (e.g., replace floor mopping solution every three patient rooms, change no less often than at 60-minute intervals), according to the facility's policy. *Category IB*. [68, 379]
f. Decontaminate mop heads and cleaning cloths regularly to prevent contamination (e.g., launder and dry at least daily). *Category II*. [68, 402, 403]
g. Use a one-step process and an EPA-registered hospital disinfectant designed for housekeeping purposes in patient care areas where 1) uncertainty exists about the nature of the soil on the surfaces (e.g., blood or body fluid contamination versus routine dust or dirt); or 2) uncertainty exists about the presence of multidrug resistant organisms on such surfaces. See 5n for recommendations requiring cleaning and disinfecting blood-contaminated surfaces. *Category II*. [23, 47, 48, 51, 214, 378, 379, 382, 416, 1012]

h. Detergent and water are adequate for cleaning surfaces in nonpatient-care areas (e.g., administrative offices). *Category II.* [23]

i. Do not use high-level disinfectants/liquid chemical sterilants for disinfection of non-critical surfaces. *Category IB*. [23, 69, 318]
j. Wet-dust horizontal surfaces regularly (e.g., daily, three times per week) using clean cloths moistened with an EPA-registered hospital disinfectant (or detergent). Prepare the disinfectant (or detergent) as recommended by the manufacturer. *Category II*. [68, 378, 380, 402, 403, 1008]
k. Disinfect noncritical surfaces with an EPA-registered hospital disinfectant according to the label's safety precautions and use directions. Most EPA-registered hospital disinfectants have a label contact time of 10 minutes. However, many scientific studies have demonstrated the efficacy of hospital disinfectants against pathogens with a contact time of at least 1 minute. By law, the user must follow all applicable label instructions on EPA-registered products. If the user selects exposure conditions that differ from those on the EPA-registered product label, the user assumes liability for any injuries resulting from off-label use and is potentially subject to enforcement action under FIFRA. *Category II, IC*. [17, 47, 48, 50, 51, 53-57, 59, 60, 62-64, 355, 378, 382]
l. Do not use disinfectants to clean infant bassinets and incubators while these items are occupied. If disinfectants (e.g., phenolics) are used for the terminal cleaning of infant bassinets and incubators, thoroughly rinse the surfaces of these items with water and dry them before these items are reused. *Category IB*. [17, 739, 740]
m. Promptly clean and decontaminate spills of blood and other potentially infectious materials. Discard blood-contaminated items in compliance with federal regulations. *Category IB, IC*. [214]
n. For site decontamination of spills of blood or other potentially infectious materials (OPIM), implement the following procedures. Use protective gloves and other PPE (e.g., when sharps are involved use forceps to pick up sharps, and discard these items in a puncture-resistant container) appropriate for this task. Disinfect areas contaminated with blood spills using an EPA-registered tuberculocidal agent, a registered germicide on the EPA Lists D and E (i.e., products with specific label claims for HIV or HBV or freshly diluted hypochlorite solution. *Category II, IC*. [214, 215, 557, 1013] If sodium hypochlorite solutions are selected use a 1:100 dilution (e.g., 1:100 dilution of a 5.25-6.15% sodium hypochlorite provides 525-615 ppm available chlorine) to decontaminate nonporous surfaces after a small spill (e.g., <10 mL) of either blood or OPIM. If a spill involves large amounts (e.g., >10 mL) of blood or OPIM, or involves a culture spill in the laboratory, use a 1:10 dilution for the first application of hypochlorite solution before cleaning in order to reduce the risk of infection during the cleaning process in the event of a sharp injury. Follow this decontamination process with a terminal disinfection, using a 1:100 dilution of sodium hypochlorite. *Category IB, IC*. [63, 215, 557]
o. If the spill contains large amounts of blood or body fluids, clean the visible matter with disposable absorbent material, and discard the contaminated materials in appropriate, labeled containment. *Category II, IC.* [44, 214]
p. Use protective gloves and other PPE appropriate for this task. *Category II, IC*. [44, 214]

q. In units with high rates of endemic *Clostridium difficile* infection or in an outbreak setting, use dilute solutions of 5.25%–6.15% sodium hypochlorite (e.g., 1:10 dilution of household bleach) for routine environmental disinfection. Currently, no products are EPA-registered specifically for inactivating *C. difficile* spores. *Category II*. [257-259]

r. If chlorine solution is not prepared fresh daily, it can be stored at room temperature for up to 30 days in a capped, opaque plastic bottle with a 50% reduction in chlorine concentration after 30 days of storage (e.g., 1000 ppm chlorine [approximately a 1:50 dilution] at day 0 decreases to 500 ppm chlorine by day 30). *Category IB*. [327, 1014]

s. An EPA-registered sodium hypochlorite product is preferred, but if such products are not available, generic versions of sodium hypochlorite solutions (e.g., household chlorine bleach) can be used. *Category II.* [44]

6. Disinfectant Fogging

a. Do not perform disinfectant fogging for routine purposes in patient-care areas. C*ategory II*. [23, 228]

7. High-Level Disinfection of Endoscopes

a. To detect damaged endoscopes, test each flexible endoscope for leaks as part of each reprocessing cycle. Remove from clinical use any instrument that fails the leak test, and repair this instrument. *Category II*. [113, 115, 116]

b. Immediately after use, meticulously clean the endoscope with an enzymatic cleaner that is compatible with the endoscope. Cleaning is necessary before both automated and manual disinfection. *Category IA*. [83, 101, 104-106, 113, 115, 116, 124, 126, 456, 465, 466, 471, 1015]

c. Disconnect and disassemble endoscopic components (e.g., suction valves) as completely as possible and completely immerse all components in the enzymatic cleaner. Steam sterilize these components if they are heat stable. *Category IB*. [115, 116, 139, 465, 466]

d. Flush and brush all accessible channels to remove all organic (e.g., blood, tissue) and other residue. Clean the external surfaces and accessories of the devices by using a soft cloth or sponge or brushes. Continue brushing until no debris appears on the brush. *Category IA* [6, 17, 108, 113, 115, 116, 137, 145, 147, 725, 856, 903].

e. Use cleaning brushes appropriate for the size of the endoscope channel or port (e.g., bristles should contact surfaces). Cleaning items (e.g., brushes, cloth) should be disposable or, if they are not disposable, they should be thoroughly cleaned and either high-level disinfected or sterilized after each use. *Category II*. [113, 115, 116, 1016]

f. Discard enzymatic cleaners (or detergents) after each use because they are not microbicidal and, therefore, will not retard microbial growth. *Category IB*. [38, 113, 115, 116, 466]

g. Process endoscopes (e.g., arthroscopes, cystoscope, laparoscopes) that pass through normally sterile tissues using a sterilization procedure before each use; if this is not feasible, provide at least high-level disinfection. High-level disinfection of arthroscopes, laparoscopes, and cytoscopes should be followed by a sterile water rinse. *Category IB*. [1, 17, 31, 32, 35, 89, 90, 113, 554]

h. Phase out endoscopes that are critical items (e.g., arthroscopes, laparoscopes) but cannot be steam sterilized. Replace these endoscopes with steam sterilizable instruments when feasible. *Category II*.

i. Mechanically clean reusable accessories inserted into endoscopes (e.g., biopsy forceps or other cutting instruments) that break the mucosal barrier (e.g., ultrasonically clean biopsy forceps) and then sterilize these items between each patient. *Category IA*. [1, 6, 8, 17, 108, 113, 115, 116, 138, 145, 147, 153, 278]

j. Use ultrasonic cleaning of reusable endoscopic accessories to remove soil and organic material from hard-to-clean areas. *Category II*. [116, 145, 148]

k. Process endoscopes and accessories that contact mucous membranes as semicritical items, and use at least high-level disinfection after use on each patient. *Category IA*. [1, 6, 8, 17, 108, 113, 115, 116, 129, 138, 145-148, 152-154, 278]

l. Use an FDA-cleared sterilant or high-level disinfectant for sterilization or high-level disinfection (Table 1). *Category IA*. [1, 6-8, 17, 85, 108, 113, 115, 116, 147]

m. After cleaning, use formulations containing glutaraldehyde, glutaraldehyde with phenol/phenate,

ortho-phthalaldehyde, hydrogen peroxide, and both hydrogen peroxide and peracetic acid to achieve high-level disinfection followed by rinsing and drying (see Table 1 for recommended concentrations). *Category IB*. [1, 6-8, 17, 38, 85, 108, 113, 145-148]

n. Extend exposure times beyond the minimum effective time for disinfecting semicritical patient-care equipment cautiously and conservatively because extended exposure to a high-level disinfectant is more likely to damage delicate and intricate instruments such as flexible endoscopes. The exposure times vary among the Food and Drug Administration (FDA)-cleared high-level disinfectants (Table 2). *Category IB*. [17, 69, 73, 76, 78, 83]

o. Federal regulations are to follow the FDA-cleared label claim for high-level disinfectants. The FDA-cleared labels for high-level disinfection with >2% glutaraldehyde at 25°C range from 20-90 minutes, depending upon the product based on three tier testing which includes AOAC sporicidal tests, simulated use testing with mycobacterial and in-use testing. *Category IC*.

p. Several scientific studies and professional organizations support the efficacy of >2% glutaraldehyde for 20 minutes at 20ºC; that efficacy assumes adequate cleaning prior to disinfection, whereas the FDA-cleared label claim incorporates an added margin of safety to accommodate possible lapses in cleaning practices. Facilities that have chosen to apply the 20 minute duration at 20ºC have done so based on the IA recommendation in the July 2003 SHEA position paper, "Multi-society Guideline for Reprocessing Flexible Gastrointestinal Endoscopes [12, 17, 19, 26, 27, 49, 55, 57, 58, 60, 73, 76, 79-81, 83-85, 93, 94, 104-106, 110, 111, 115-121, 124, 125, 233, 235, 236, 243, 265, 266, 609]

q. When using FDA-cleared high-level disinfectants, use manufacturers' recommended exposure conditions. Certain products may require a shorter exposure time (e.g., 0.55% ortho-phthalaldehyde for 12 minutes at 20°C, 7.35% hydrogen peroxide plus 0.23% peracetic acid for 15 minutes at 20°C) than glutaraldehyde at room temperature because of their rapid inactivation of mycobacteria or reduced exposure time because of increased mycobactericidal activity at elevated temperature (e.g., 2.5% glutaraldehyde at 5 minutes at 35°C). *Category IB*. [83, 100, 689, 693, 694, 700]

r. Select a disinfectant or chemical sterilant that is compatible with the device that is being reprocessed. Avoid using reprocessing chemicals on an endoscope if the endoscope manufacturer warns against using these chemicals because of functional damage (with or without cosmetic damage). *Category IB*. [69, 113, 116]

s. Completely immerse the endoscope in the high-level disinfectant, and ensure all channels are perfused. As soon as is feasible, phase out nonimmersible endoscopes. *Category IB*. [108, 113-116, 137, 725, 856, 882]

t. After high-level disinfection, rinse endoscopes and flush channels with sterile water, filtered water, or tapwater to prevent adverse effects on patients associated with disinfectant retained in the endoscope (e.g., disinfectant induced colitis). Follow this water rinse with a rinse with 70% - 90% ethyl or isopropyl alcohol. *Category IB*. [17, 31-35, 38, 39, 108, 113, 115, 116, 134, 145-148, 620-622, 624-630, 1017]

u. After flushing all channels with alcohol, purge the channels using forced air to reduce the likelihood of contamination of the endoscope by waterborne pathogens and to facilitate drying. *Category IB*. [39, 113, 115, 116, 145, 147]

v. Hang endoscopes in a vertical position to facilitate drying. *Category II*. [17, 108, 113, 115, 116, 145, 815]

w. Store endoscopes in a manner that will protect them from damage or contamination. *Category II*. [17, 108, 113, 115, 116, 145]

x. Sterilize or high-level disinfect both the water bottle used to provide intraprocedural flush solution and its connecting tube at least once daily. After sterilizing or high-level disinfecting the water bottle, fill it with sterile water. *Category IB*. [10, 31-35, 113, 116, 1017]

y. Maintain a log for each procedure and record the following: patient's name and medical record number (if available), procedure, date, endoscopist, system used to reprocess the endoscope (if more than one system could be used in the reprocessing area), and serial number or other identifier of the endoscope used. *Category II*. [108, 113, 115, 116]

z. Design facilities where endoscopes are used and disinfected to provide a safe environment for healthcare workers and patients. Use air-exchange equipment (e.g., the ventilation system, out-exhaust ducts) to minimize exposure of all persons to potentially toxic vapors (e.g.,

glutaraldehyde vapor). Do not exceed the allowable limits of the vapor concentration of the chemical sterilant or high-level disinfectant (e.g., those of ACGIH and OSHA). *Category IB, IC*. [116, 145, 318, 322, 577, 652]

aa. Routinely test the liquid sterilant/high-level disinfectant to ensure minimal effective concentration of the active ingredient. Check the solution each day of use (or more frequently) using the appropriate chemical indicator (e.g., glutaraldehyde chemical indicator to test minimal effective concentration of glutaraldehyde) and document the results of this testing. Discard the solution if the chemical indicator shows the concentration is less than the minimum effective concentration. Do not use the liquid sterilant/high-level disinfectant beyond the reuse-life recommended by the manufacturer (e.g., 14 days for *ortho*-phthalaldehyde). *Category IA*. [76, 108, 113, 115, 116, 608, 609]

bb. Provide personnel assigned to reprocess endoscopes with device-specific reprocessing instructions to ensure proper cleaning and high-level disinfection or sterilization. Require competency testing on a regular basis (e.g., beginning of employment, annually) of all personnel who reprocess endoscopes. *Category IA*. [6-8, 108, 113, 115, 116, 145, 148, 155]

cc. Educate all personnel who use chemicals about the possible biologic, chemical, and environmental hazards of performing procedures that require disinfectants. C*ategory IB, IC*. [116, 997, 998, 1018, 1019]

dd. Make PPE(e.g., gloves, gowns, eyewear, face mask or shields, respiratory protection devices) available and use these items appropriately to protect workers from exposure to both chemicals and microorganisms (e.g., HBV). *Category IB, IC*. [115, 116, 214, 961, 997, 998, 1020, 1021]

ee. If using an automated endoscope reprocessor (AER), place the endoscope in the reprocessor and attach all channel connectors according to the AER manufacturer's instructions to ensure exposure of all internal surfaces to the high-level disinfectant/chemical sterilant. *Category IB*. [7, 8, 115, 116, 155, 725, 903]

ff. If using an AER, ensure the endoscope can be effectively reprocessed in the AER. Also, ensure any required manual cleaning/disinfecting steps are performed (e.g., elevator wire channel of duodenoscopes might not be effectively disinfected by most AERs). *Category IB*. [7, 8, 115, 116, 155, 725]

gg. Review the FDA advisories and the scientific literature for reports of deficiencies that can lead to infection because design flaws and improper operation and practices have compromised the effectiveness of AERs. *Category II*. [7, 98, 133, 134, 155, 725]

hh. Develop protocols to ensure that users can readily identify an endoscope that has been properly processed and is ready for patient use. *Category II*.

ii. Do not use the carrying case designed to transport clean and reprocessed endoscopes outside of the healthcare environment to store an endoscope or to transport the instrument within the healthcare environment. *Category II.*

jj. No recommendation is made about routinely performing microbiologic testing of either endoscopes or rinse water for quality assurance purposes. *Unresolved Issue.* [116, 164]

kk. If environmental microbiologic testing is conducted, use standard microbiologic techniques*. Category II.* [23, 116, 157, 161, 167]

ll. If a cluster of endoscopy-related infections occurs, investigate potential routes of transmission (e.g., person-to-person, common source) and reservoirs. *Category IA*. [8, 1022]

mm. Report outbreaks of endoscope-related infections to persons responsible for institutional infection control and risk management and to FDA. *Category IB*. [6, 7, 113, 116, 1023] Notify the local and the state health departments, CDC, and the manufacturer(s). C*ategory II*.

nn. No recommendation is made regarding the reprocessing of an endoscope again immediately before use if that endoscope has been processed after use according to the recommendations in this guideline. *Unresolved issue*. [157]

oo. Compare the reprocessing instructions provided by both the endoscope's and the AER's manufacturer's instructions and resolve any conflicting recommendations. *Category IB*. [116, 155]

8. *Management of Equipment and Surfaces in Dentistry*

a. Dental instruments that penetrate soft tissue or bone (e.g., extraction forceps, scalpel blades, bone chisels, periodontal scalers, and surgical burs) are classified as critical and should be

sterilized after each use or discarded. In addition, after each use, sterilize dental instruments that are not intended to penetrate oral soft tissue or bone (e.g., amalgam condensers, air-water syringes) but that might contact oral tissues and are heat-tolerant, although classified as semicritical. Clean and, at a minimum, high-level disinfect heat-sensitive semicritical items. *Category IA*. [43, 209-211]

b. Noncritical clinical contact surfaces, such as uncovered operatory surfaces (e.g., countertops, switches, light handles), should be barrier-protected or disinfected between patients with an intermediate-disinfectant (i.e., EPA-registered hospital disinfectant with a tuberculocidal claim) or low-level disinfectant (i.e., EPA-registered hospital disinfectant with HIV and HBV claim). *Category IB*. [43, 209-211]

c. Barrier protective coverings can be used for noncritical clinical contact surfaces that are touched frequently with gloved hands during the delivery of patient care, that are likely to become contaminated with blood or body substances, or that are difficult to clean. Change these coverings when they are visibly soiled, when they become damaged, and on a routine basis (e.g., between patients). Disinfect protected surfaces at the end of the day or if visibly soiled. *Category II*. [43, 210]

9. Processing Patient-Care Equipment Contaminated with Bloodborne Pathogens (HBV, Hepatitis C Virus, HIV), Antibiotic-Resistant Bacteria (e.g., Vancomycin-Resistant Enterococci, Methicillin-Resistant Staphylococcus aureus, Multidrug Resistant Tuberculosis), or Emerging Pathogens (e.g., Cryptosporidium, Helicobacter pylori, Escherichia coli O157:H7, Clostridium difficile, Mycobacterium tuberculosis, Severe Acute Respiratory Syndrome Coronavirus), or Bioterrorist Agents

a. Use standard sterilization and disinfection procedures for patient-care equipment (as recommended in this guideline), because these procedures are adequate to sterilize or disinfect instruments or devices contaminated with blood or other body fluids from persons infected with bloodborne pathogens or emerging pathogens, with the exception of prions. No changes in these procedures for cleaning, disinfecting, or sterilizing are necessary for removing bloodborne and emerging pathogens other than prions. *Category IA*. [22, 53, 60-62, 73, 79-81, 105, 118-121, 125, 126, 221, 224-234, 236, 244, 265, 266, 271-273, 279, 282, 283, 354-357, 666]

10. Disinfection Strategies for Other Semicritical Devices

a. Even if probe covers have been used, clean and high-level disinfect other semicritical devices such as rectal probes, vaginal probes, and cryosurgical probes with a product that is not toxic to staff, patients, probes, and retrieved germ cells (if applicable). Use a high-level disinfectant at the FDA-cleared exposure time. (See Recommendations 7o and 11e for exceptions.) *Category IB*. [6-8, 17, 69]

b. When probe covers are available, use a probe cover or condom to reduce the level of microbial contamination. *Category II*. [197-201] Do not use a lower category of disinfection or cease to follow the appropriate disinfectant recommendations when using probe covers because these sheaths and condoms can fail. *Category IB* [197-201]

c. After high-level disinfection, rinse all items. Use sterile water, filtered water or tapwater followed by an alcohol rinse for semicritical equipment that will have contact with mucous membranes of the upper respiratory tract (e.g., nose, pharynx, esophagus). *Category II*. [10, 31-35, 1017]

d. There is no recommendation to use sterile or filtered water rather than tapwater for rinsing semicritical equipment that contact the mucous membranes of the rectum (e.g., rectal probes, anoscope) or vagina (e.g., vaginal probes). *Unresolved issue.* [11]

e. Wipe clean tonometer tips and then disinfect them by immersing for 5-10 minutes in either 5000 ppm chlorine or 70% ethyl alcohol. None of these listed disinfectant products are FDA-cleared high-level disinfectants. *Category II*. [49, 95, 185, 188, 293]

11. Disinfection by Healthcare Personnel in Ambulatory Care and Home Care

a. Follow the same classification scheme described above (i.e., that critical devices require sterilization, semicritical devices require high-level disinfection, and noncritical equipment

requires low-level disinfection) in the ambulatory-care (outpatient medical/surgical facilities) setting because risk for infection in this setting is similar to that in the hospital setting (see Table 1). *Category IB*. [6-8, 17, 330]

b. When performing care in the home, clean and disinfect reusable objects that touch mucous membranes (e.g., tracheostomy tubes) by immersing these objects in a 1:50 dilution of 5.25%-6.15% sodium hypochlorite (household bleach) (3 minutes), 70% isopropyl alcohol (5 minutes), or 3% hydrogen peroxide (30 minutes) because the home environment is, in most instances, safer than either hospital or ambulatory care settings because person-to-person transmission is less likely. *Category II*. [327, 328, 330, 331]

c. Clean noncritical items that would not be shared between patients (e.g., crutches, blood pressure cuffs) in the home setting with a detergent or commercial household disinfectant. *Category II*. [53, 330]

12. Microbial Contamination of Disinfectants

a. Institute the following control measures to reduce the occurrence of contaminated disinfectants: 1) prepare the disinfectant correctly to achieve the manufacturer's recommended use-dilution; and 2) prevent common sources of extrinsic contamination of germicides (e.g., container contamination or surface contamination of the healthcare environment where the germicide are prepared and/or used). *Category IB*. [404, 406, 1024]

13. Flash Sterilization

a. Do not flash sterilize implanted surgical devices unless doing so is unavoidable. *Category IB*. [849, 850]

b. Do not use flash sterilization for convenience, as an alternative to purchasing additional instrument sets, or to save time. Category II. [817, 962]

c. When using flash sterilization, make sure the following parameters are met: 1) clean the item before placing it in the sterilizing container (that are FDA cleared for use with flash sterilization) or tray; 2) prevent exogenous contamination of the item during transport from the sterilizer to the patient; and 3) monitor sterilizer function with mechanical, chemical, and biologic monitors. *Category IB*. [812, 819, 846, 847, 962]

d. Do not use packaging materials and containers in flash sterilization cycles unless the sterilizer and the packaging material/container are designed for this use. *Category IB*. [812, 819, 1025]

e. When necessary, use flash sterilization for patient-care items that will be used immediately (e.g., to reprocess an inadvertently dropped instrument). *Category IB*. [812, 817, 819, 845]

f. When necessary, use flash sterilization for processing patient-care items that cannot be packaged, sterilized, and stored before use. *Category IB*. [812, 819]

14. Methods of Sterilization

a. Steam is the preferred method for sterilizing critical medical and surgical instruments that are not damaged by heat, steam, pressure, or moisture. *Category IA*. [181, 271, 425, 426, 827, 841, 1026, 1027]

b. Cool steam- or heat-sterilized items before they are handled or used in the operative setting. *Category IB*. [850]

c. Follow the sterilization times, temperatures, and other operating parameters (e.g., gas concentration, humidity) recommended by the manufacturers of the instruments, the sterilizer, and the container or wrap used, and that are consistent with guidelines published by government agencies and professional organizations. *Category IB*. [811-814, 819, 825, 827, 841, 1026-1028]

d. Use low-temperature sterilization technologies (e.g., EtO, hydrogen peroxide gas plasma) for reprocessing critical patient-care equipment that is heat or moisture sensitive. *Category IA* [469, 721, 825, 856, 858, 878, 879, 881, 882, 890, 891, 1027].

e. Completely aerate surgical and medical items that have been sterilized in the EtO sterilizer (e.g., polyvinylchloride tubing requires 12 hours at 50°C, 8 hours at 60°C) before using these items in patient care. *Category IB*. [814]

f. Sterilization using the peracetic acid immersion system can be used to sterilize heat-sensitive

immersible medical and surgical items. *Category IB*. [90, 717-719, 721-724]

g. Critical items that have been sterilized by the peracetic acid immersion process must be used immediately (i.e., items are not completely protected from contamination, making long-term storage unacceptable). *Category II*. [817, 825]
h. Dry-heat sterilization (e.g., 340°F for 60 minutes) can be used to sterilize items (e.g., powders, oils) that can sustain high temperatures. *Category IB*. [815, 827]
i. Comply with the sterilizer manufacturer's instructions regarding the sterilizer cycle parameters (e.g., time, temperature, concentration). *Category IB*. [155, 725, 811-814, 819]
j. Because narrow-lumen devices provide a challenge to all low-temperature sterilization technologies and direct contact is necessary for the sterilant to be effective, ensure that the sterilant has direct contact with contaminated surfaces (e.g., scopes processed in peracetic acid must be connected to channel irrigators). *Category IB*. [137, 725, 825, 856, 890, 891, 1029]

15. Packaging

a. Ensure that packaging materials are compatible with the sterilization process and have received FDA 510[k] clearance. *Category IB*. [811-814, 819, 966]
b. Ensure that packaging is sufficiently strong to resist punctures and tears to provide a barrier to microorganisms and moisture. *Category IB*. [454, 811-814, 819, 966]

16. Monitoring of Sterilizers

a. Use mechanical, chemical, and biologic monitors to ensure the effectiveness of the sterilization process. *Category IB*. [811-815, 819, 846, 847, 975-977]
b. Monitor each load with mechanical (e.g., time, temperature, pressure) and chemical (internal and external) indicators. If the internal chemical indicator is visible, an external indicator is not needed. *Category II*. [811-815, 819, 846, 847, 975-977, 980]
c. Do not use processed items if the mechanical (e.g., time, temperature, pressure) or chemical (internal and/or external) indicators suggest inadequate processing. *Category IB* [811-814, 819].
d. Use biologic indicators to monitor the effectiveness of sterilizers at least weekly with an FDA-cleared commercial preparation of spores (e.g., *Geobacillus stearothermophilus* for steam) intended specifically for the type and cycle parameters of the sterilizer. *Category IB*. [1, 811, 813-815, 819, 846, 847, 976, 977]
e. After a single positive biologic indicator used with a method other than steam sterilization, treat as nonsterile all items that have been processed in that sterilizer, dating from the sterilization cycle having the last negative biologic indicator to the next cycle showing satisfactory biologic indicator results. These nonsterile items should be retrieved if possible and reprocessed. *Category II*. [1]
f. After a positive biologic indicator with steam sterilization, objects other than implantable objects do not need to be recalled because of a single positive spore test unless the sterilizer or the sterilization procedure is defective as determined by maintenance personnel or inappropriate cycle settings. If additional spore tests remain positive, consider the items nonsterile and recall and reprocess the items from the implicated load(s). *Category II*. [1]
g. Use biologic indicators for every load containing implantable items and quarantine items, whenever possible, until the biologic indicator is negative. *Category IB*. [811-814, 819]

17. Load Configuration.

a. Place items correctly and loosely into the basket, shelf, or cart of the sterilizer so as not to impede the penetration of the sterilant. *Category IB*. [445, 454, 811, 813, 819, 836]

18. Storage of Sterile Items

a. Ensure the sterile storage area is a well-ventilated area that provides protection against dust, moisture, insects, and temperature and humidity extremes. *Category II*. [454, 819, 836, 969]
b. Store sterile items so the packaging is not compromised (e.g., punctured, bent). *Category II*. [454, 816, 819, 968, 969, 1030]

c. Label sterilized items with a load number that indicates the sterilizer used, the cycle or load number, the date of sterilization, and, if applicable, the expiration date. *Category IB*. [811, 812, 814, 816, 819]

d. The shelf life of a packaged sterile item depends on the quality of the wrapper, the storage conditions, the conditions during transport, the amount of handling, and other events (moisture) that compromise the integrity of the package. If event-related storage of sterile items is used, then packaged sterile items can be used indefinitely unless the packaging is compromised (see f and g below). *Category IB*. [816, 819, 836, 968, 973, 1030, 1031]

e. Evaluate packages before use for loss of integrity (e.g., torn, wet, punctured). The pack can be used unless the integrity of the packaging is compromised. *Category II*. [819, 968]

f. If the integrity of the packaging is compromised (e.g., torn, wet, or punctured), repack and reprocess the pack before use. *Category II*. [819, 1032]

g. If time-related storage of sterile items is used, label the pack at the time of sterilization with an expiration date. Once this date expires, reprocess the pack. *Category II*. [819, 968]

19. Quality Control

a. Provide comprehensive and intensive training for all staff assigned to reprocess semicritical and critical medical/surgical instruments to ensure they understand the importance of reprocessing these instruments. To achieve and maintain competency, train each member of the staff that reprocesses semicritical and/or critical instruments as follows: 1) provide hands-on training according to the institutional policy for reprocessing critical and semicritical devices; 2) supervise all work until competency is documented for each reprocessing task; 3) conduct competency testing at beginning of employment and regularly thereafter (e.g., annually); and 4) review the written reprocessing instructions regularly to ensure they comply with the scientific literature and the manufacturers' instructions. *Category IB*. [6-8, 108, 114, 129, 155, 725, 813, 819]

b. Compare the reprocessing instructions (e.g., for the appropriate use of endoscope connectors, the capping/noncapping of specific lumens) provided by the instrument manufacturer and the sterilizer manufacturer and resolve any conflicting recommendations by communicating with both manufacturers. *Category IB*. [155, 725]

c. Conduct infection control rounds periodically (e.g., annually) in high-risk reprocessing areas (e.g., the Gastroenterology Clinic, Central Processing); ensure reprocessing instructions are current and accurate and are correctly implemented. Document all deviations from policy. All stakeholders should identify what corrective actions will be implemented. *Category IB*. [6-8, 129]

d. Include the following in a quality control program for sterilized items: a sterilizer maintenance contract with records of service; a system of process monitoring; air-removal testing for prevacuum steam sterilizers; visual inspection of packaging materials; and traceability of load contents. *Category II* [811-814, 819].

e. For each sterilization cycle, record the type of sterilizer and cycle used; the load identification number; the load contents; the exposure parameters (e.g., time and temperature); the operator's name or initials; and the results of mechanical, chemical, and biological monitoring. *Category II* [811-814, 819].

f. Retain sterilization records (mechanical, chemical, and biological) for a time period that complies with standards (e.g., 3 years), statutes of limitations, and state and federal regulations. *Category II, IC*. [1033]

g. Prepare and package items to be sterilized so that sterility can be achieved and maintained to the point of use. Consult the Association for the Advancement of Medical Instrumentation or the manufacturers of surgical instruments, sterilizers, and container systems for guidelines for the density of wrapped packages. *Category II*. [811-814, 819]

h. Periodically review policies and procedures for sterilization. *Category II*. [1033]

i. Perform preventive maintenance on sterilizers by qualified personnel who are guided by the manufacturer's instruction. *Category II*. [811-814, 819]

20. Reuse of Single-Use Medical Devices

a. Adhere to the FDA enforcement document for single-use devices reprocessed by hospitals. FDA considers the hospital that reprocesses a single-use device as the manufacturer of the device and regulates the hospital using the same standards by which it regulates the original equipment manufacturer. *Category II, IC*. [995]

PERFORMANCE INDICATORS

1. Monitor adherence to high-level disinfection and/or sterilization guidelines for endoscopes on a regular basis. This monitoring should include ensuring the proper training of persons performing reprocessing and their adherence to all endoscope reprocessing steps, as demonstrated by competency testing at commencement of employment and annually.
2. Develop a mechanism for the occupational health service to report all adverse health events potentially resulting from exposure to disinfectants and sterilants; review such exposures; and implement engineering, work practice, and PPE to prevent future exposures.
3. Monitor possible sterilization failures that resulted in instrument recall. Assess whether additional training of personnel or equipment maintenance is required.

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GLOSSARY

Action level: concentration of a regulated substance (e.g., ethylene oxide, formaldehyde) within the employee breathing zone, above which OSHA requirements apply.

Activation of a sterilant: process of mixing the contents of a chemical sterilant that come in two containers (small vial with the activator solution; container of the chemical) Keeping the two chemicals separate until use extends the shelf life of the chemicals.

Aeration: method by which ethylene oxide (EtO) is removed from EtO-sterilized items by warm air circulation in an enclosed cabinet specifically designed for this purpose.

Antimicrobial agent: any agent that kills or suppresses the growth of microorganisms.

Antiseptic: substance that prevents or arrests the growth or action of microorganisms by inhibiting their activity or by destroying them. The term is used especially for preparations applied topically to living tissue.

Asepsis: prevention of contact with microorganisms.

Autoclave: device that sterilizes instruments or other objects using steam under pressure. The length of time required for sterilization depends on temperature, vacuum, and pressure.

Bacterial count: method of estimating the number of bacteria per unit sample. The term also refers to the estimated number of bacteria per unit sample, usually expressed as number of colony-forming units.

Bactericide: agent that kills bacteria.

Bioburden: number and types of viable microorganisms with which an item is contaminated; also called *bioload* or *microbial load*.

Biofilm: accumulated mass of bacteria and extracellular material that is tightly adhered to a surface and cannot be easily removed.

Biologic indicator: device for monitoring the sterilization process. The device consists of a standardized, viable population of microorganisms (usually bacterial spores) known to be resistant to the sterilization process being monitored. Biologic indicators are intended to demonstrate whether conditions were adequate to achieve sterilization. A negative biologic indicator does not prove that all items in the load are sterile or that they were all exposed to adequate sterilization conditions.

Bleach: Household bleach (5.25% or 6.00%–6.15% sodium hypochlorite depending on manufacturer) usually diluted in water at 1:10 or 1:100. Approximate dilutions are 1.5 cups of bleach in a gallon of water for a 1:10 dilution (~6,000 ppm) and 0.25 cup of bleach in a gallon of water for a 1:100 dilution (~600 ppm). Sodium hypochlorite products that make pesticidal claims, such as sanitization or disinfection, must be registered by EPA and be labeled with an EPA Registration Number.

Bleach Solution	Dilution	Chlorine (ppm)
5.25-6.15%	None	52,500-61,500
	1:10	5,250-6,150
	1:100	525-615
	1:1000	53-62

Bowie-Dick test: diagnostic test of a sterilizer's ability to remove air from the chamber of a prevacuum steam sterilizer. The air-removal or Bowie-Dick test is not a test for sterilization.

Ceiling limit: concentration of an airborne chemical contaminant that should not be exceeded during any part of the workday. If instantaneous monitoring is not feasible, the ceiling must be assessed as a 15-minute time-weighted average exposure.

Centigrade or **Celsius**: a temperature scale ($0^{o}C$ = freezing point of water; $100^{o}C$ = boiling point of water at sea level). Equivalents mentioned in the guideline are as follows: $20^{o}C = 68^{o}F$; $25^{o}C = 77^{o}F$; $121^{o}C = 250^{o}F$; $132^{o}C = 270^{o}F$; $134^{o}C = 273^{o}F$. For other temperatures the formula is: $F^{o} = (C^{o} \times 9/5) + 32$ *or* $C^{o} = (F^{o} - 32) \times 5/9$.

Central processing or **Central service department**: the department within a health-care facility that processes, issues, and controls professional supplies and equipment, both sterile and nonsterile, for some or all patient-care areas of the facility.

Challenge test pack: pack used in installation, qualification, and ongoing quality assurance testing of health-care facility sterilizers.

Chemical indicator: device for monitoring a sterilization process. The device is designed to respond with a characteristic chemical or physical change to one or more of the physical conditions within the sterilizing chamber. Chemical indicators are intended to detect potential sterilization failures that could result from incorrect packaging, incorrect loading of the sterilizer, or malfunctions of the sterilizer. The "pass" response of a chemical indicator does not prove the item accompanied by the indicator is necessarily sterile. The Association for the Advancement of Medical Instrumentation has defined five classes of chemical indicators: Class 1 (process indicator); Class 2 (Bowie-Dick test indicator); Class 3 (single-parameter indicator); Class 4 (multi-parameter indicator); and Class 5 (integrating indicator).

Contact time: time a disinfectant is in direct contact with the surface or item to be disinfected For surface disinfection, this period is framed by the application to the surface until complete drying has occurred.

Container system, rigid container: sterilization containment device designed to hold medical devices for sterilization, storage, transportation, and aseptic presentation of contents.

Contaminated: state of having actual or potential contact with microorganisms. As used in health care, the term generally refers to the presence of microorganisms that could produce disease or infection.

Control, positive: biologic indicator, from the same lot as a test biologic indicator, that is left unexposed to the sterilization cycle and then incubated to verify the viability of the test biologic indicator.

Cleaning: removal, usually with detergent and water or enzyme cleaner and water, of adherent visible soil, blood, protein substances, microorganisms and other debris from the surfaces, crevices, serrations, joints, and lumens of instruments, devices, and equipment by a manual or mechanical process that prepares the items for safe handling and/or further decontamination.

Culture: growth of microorganisms in or on a nutrient medium; to grow microorganisms in or on such a medium.

Culture medium: substance or preparation used to grow and cultivate microorganisms.

Cup: 8 fluid ounces.

Decontamination: according to OSHA, "the use of physical or chemical means to remove, inactivate, or destroy bloodborne pathogens on a surface or item to the point where they are no longer capable of transmitting infectious particles and the surface or item is rendered safe for handling, use, or disposal" [29 CFR 1910.1030]. In health-care facilities, the term generally refers to all pathogenic organisms.

Decontamination area: area of a health-care facility designated for collection, retention, and cleaning of soiled and/or contaminated items.

Detergent: cleaning agent that makes no antimicrobial claims on the label. They comprise a hydrophilic component and a lipohilic component and can be divided into four types: anionic, cationic, amphoteric, and non-ionic detergents.

Disinfectant: usually a chemical agent (but sometimes a physical agent) that destroys disease-causing pathogens or other harmful microorganisms but might not kill bacterial spores. It refers to substances applied to inanimate objects. EPA groups disinfectants by product label claims of "limited," "general," or "hospital" disinfection.

Disinfection: thermal or chemical destruction of pathogenic and other types of microorganisms. Disinfection is less lethal than sterilization because it destroys most recognized pathogenic microorganisms but not necessarily all microbial forms (e.g., bacterial spores).

D value: time or radiation dose required to inactivate 90% of a population of the test microorganism under stated exposure conditions.

Endoscope: an instrument that allows examination and treatment of the interior of the body canals and hollow organs.

Enzyme cleaner: a solution used before disinfecting instruments to improve removal of organic material (e.g., proteases to assist in removing protein).

EPA Registration Number or **EPA Reg. No.**: a hyphenated, two- or three-part number assigned by EPA to identify each germicidal product registered within the United States. The first number is the company identification number, the second is the specific product number, and the third (when present) is the company identification number for a supplemental registrant.

Exposure time: period in a sterilization process during which items are exposed to the sterilant at the specified sterilization parameters. For example, in a steam sterilization process, exposure time is the period during which items are exposed to saturated steam at the specified temperature.

Flash sterilization: process designed for the steam sterilization of unwrapped patient-care items for immediate use (or placed in a specially designed, covered, rigid container to allow for rapid penetration of steam).

Fungicide: agent that destroys fungi (including yeasts) and/or fungal spores pathogenic to humans or other animals in the inanimate environment.

General disinfectant: EPA-registered disinfectant labeled for use against both gram-negative and gram-positive bacteria. Efficacy is demonstrated against both *Salmonella choleraesuis* and *Staphylococcus aureus*. Also called *broad-spectrum disinfectant*.

Germicide: agent that destroys microorganisms, especially pathogenic organisms.

Germicidal detergent: detergent that also is EPA-registered as a disinfectant.

High-level disinfectant: agent capable of killing bacterial spores when used in sufficient concentration under suitable conditions. It therefore is expected to kill all other microorganisms.

Hospital disinfectant: disinfectant registered for use in hospitals, clinics, dental offices, and any other medical-related facility. Efficacy is demonstrated against *Salmonella choleraesuis*, *Staphylococcus aureus*, and *Pseudomonas aeruginosa.* EPA has registered approximately 1,200 hospital disinfectants**.**

Huck towel: all-cotton surgical towel with a honey-comb weave; both warp and fill yarns are tightly twisted. Huck towels can be used to prepare biologic indicator challenge test packs.

Implantable device: according to FDA, "device that is placed into a surgically or naturally formed cavity of the human body if it is intended to remain there for a period of 30 days or more" [21 CFR 812.3(d)].

Inanimate surface: nonliving surface (e.g., floors, walls, furniture).

Incubator: apparatus for maintaining a constant and suitable temperature for the growth and cultivation of microorganisms.

Infectious microorganisms: microorganisms capable of producing disease in appropriate hosts.

Inorganic and organic load: naturally occurring or artificially placed inorganic (e.g., metal salts) or organic (e.g., proteins) contaminants on a medical device before exposure to a microbicidal process.

Intermediate-level disinfectant: agent that destroys all vegetative bacteria, including tubercle bacilli, lipid and some nonlipid viruses, and fungi, but not bacterial spores.

Limited disinfectant: disinfectant registered for use against a specific major group of organisms (gram-negative or gram-positive bacteria). Efficacy has been demonstrated in laboratory tests against either *Salmonella choleraesuis* or *Staphylococcus aureus* bacteria.

Lipid virus: virus surrounded by an envelope of lipoprotein in addition to the usual core of nucleic acid surrounded by a coat of protein. This type of virus (e.g., HIV) is generally easily inactivated by many types of disinfectants. Also called *enveloped* or *lipophilic virus*.

Low-level disinfectant: agent that destroys all vegetative bacteria (except tubercle bacilli), lipid viruses, some nonlipid viruses, and some fungi, but not bacterial spores.

Mechanical indicator: devices that monitor the sterilization process (e.g., graphs, gauges, printouts).

Medical device: instrument, apparatus, material, or other article, whether used alone or in combination, including software necessary for its application, intended by the manufacturer to be used for human beings for

- diagnosis, prevention, monitoring treatment, or alleviation of disease;
- diagnosis, monitoring, treatment, or alleviation of or compensation for an injury or handicap;
- investigation, replacement, or modification of the anatomy or of a physiologic process; or
- control of conception

and that does not achieve its primary intended action in or on the human body by pharmacologic, immunologic, or metabolic means but might be assisted in its function by such means.

Microbicide: any substance or mixture of substances that effectively kills microorganisms.

Microorganisms: animals or plants of microscopic size. As used in health care, generally refers to bacteria, fungi, viruses, and bacterial spores.

Minimum effective concentration (MEC): the minimum concentration of a liquid chemical germicide needed to achieve the claimed microbicidal activity as determined by dose-response testing. Sometimes used interchangeably with *minimum recommended concentration*.

Muslin: loosely woven (by convention, 140 threads per square inch), 100% cotton cloth. Formerly used as a wrap for sterile packs or a surgical drape. Fabric wraps used currently consist of a cotton-polyester blend.

Mycobacteria: bacteria with a thick, waxy coat that makes them more resistant to chemical germicides than other types of vegetative bacteria.

Nonlipid viruses: generally considered more resistant to inactivation than lipid viruses. Also called nonenveloped or hydrophilic viruses.

One-step disinfection process: simultaneous cleaning and disinfection of a noncritical surface or item.

Pasteurization: process developed by Louis Pasteur of heating milk, wine, or other liquids to 65–77^{o}C (or the equivalent) for approximately 30 minutes to kill or markedly reduce the number of pathogenic and spoilage organisms other than bacterial spores.

Parametric release: declaration that a product is sterile on the basis of physical and/or chemical process data rather than on sample testing or biologic indicator results.

Penicylinder: carriers inoculated with the test bacteria for in vitro tests of germicides. Can be constructed of stainless steel, porcelain, glass, or other materials and are approximately 8 x 10 mm in diameter.

Permissible exposure limit (PEL): time-weighted average maximum concentration of an air contaminant to which a worker can be exposed, according to OSHA standards. Usually calculated over 8 hours, with exposure considered over a 40-hour work week.

Personal protective equipment (PPE): specialized clothing or equipment worn by an employee for protection against a hazard. General work clothes (e.g., uniforms, pants, shirts) not intended to function as protection against a hazard are not considered to be PPE.

Parts per million (ppm): common measurement for concentrations by volume of trace contaminant gases in the air (or chemicals in a liquid); 1 volume of contaminated gas per 1 million volumes of contaminated air or 1¢ in $10,000 both equal 1 ppm. Parts per million = µg/mL or mg/L.

Prions: transmissible pathogenic agents that cause a variety of neurodegenerative diseases of humans and animals, including sheep and goats, bovine spongiform encephalopathy in cattle, and Creutzfeldt-Jakob disease in humans. They are unlike any other infectious pathogens because they are composed of an abnormal conformational isoform of a normal cellular protein, the prion protein (PrP). Prions are extremely resistant to inactivation by sterilization processes and disinfecting agents.

Process challenge device (PCD): item designed to simulate product to be sterilized and to constitute a defined challenge to the sterilization process and used to assess the effective performance of the process. A PCD is a challenge test pack or test tray that contains a biologic indicator, a Class 5 integrating indicator, or an enzyme-only indicator.

QUAT: abbreviation for *quaternary ammonium compound*, a surface-active, water-soluble disinfecting

substance that has four carbon atoms linked to a nitrogen atom through covalent bonds.

Recommended exposure limit (REL): occupational exposure limit recommended by NIOSH as being protective of worker health and safety over a working lifetime. Frequently expressed as a 40-hour time-weighted-average exposure for up to 10 hours per day during a 40-work week.

Reprocess: method to ensure proper disinfection or sterilization; can include: cleaning, inspection, wrapping, sterilizing, and storing.

Sanitizer: agent that reduces the number of bacterial contaminants to safe levels as judged by public health requirements. Commonly used with substances applied to inanimate objects. According to the protocol for the official sanitizer test, a sanitizer is a chemical that kills 99.999% of the specific test bacteria in 30 seconds under the conditions of the test.

Shelf life: length of time an undiluted or use dilution of a product can remain active and effective. Also refers to the length of time a sterilized product (e.g., sterile instrument set) is expected to remain sterile.

Spaulding classification: strategy for reprocessing contaminated medical devices. The system classifies a medical device as critical, semicritical, or noncritical on the basis of risk to patient safety from contamination on a device. The system also established three levels of germicidal activity (sterilization, high-level disinfection, and low-level disinfection) for strategies with the three classes of medical devices (critical, semicritical, and noncritical).

Spore: relatively water-poor round or elliptical resting cell consisting of condensed cytoplasm and nucleus surrounded by an impervious cell wall or coat. Spores are relatively resistant to disinfectant and sterilant activity and drying conditions (specifically in the genera *Bacillus* and *Clostridium*).

Spore strip: paper strip impregnated with a known population of spores that meets the definition of biological indicators.

Steam quality: steam characteristic reflecting the dryness fraction (weight of dry steam in a mixture of dry saturated steam and entrained water) and the level of noncondensable gas (air or other gas that will not condense under the conditions of temperature and pressure used during the sterilization process). The dryness fraction (i.e., the proportion of completely dry steam in the steam being considered) should not fall below 97%.

Steam sterilization: sterilization process that uses saturated steam under pressure for a specified exposure time and at a specified temperature, as the sterilizing agent.

Steam sterilization, dynamic air removal type: one of two types of sterilization cycles in which air is removed from the chamber and the load by a series of pressure and vacuum excursions (prevacuum cycle) or by a series of steam flushes and pressure pulses above atmospheric pressure (steam-flush-pressure-pulse cycle).

Sterile or **Sterility**: state of being free from all living microorganisms. In practice, usually described as a probability function, e.g., as the probability of a microorganism surviving sterilization being one in one million.

Sterility assurance level (SAL): probability of a viable microorganism being present on a product unit after sterilization. Usually expressed as 10^{-6}; a SAL of 10^{-6} means $\leq$1/1 million chance that a single viable microorganism is present on a sterilized item. A SAL of 10^{-6} generally is accepted as appropriate for items intended to contact compromised tissue (i.e., tissue that has lost the integrity of the natural body barriers). The sterilizer manufacturer is responsible for ensuring the sterilizer can achieve the desired SAL. The

user is responsible for monitoring the performance of the sterilizer to ensure it is operating in conformance to the manufacturer's recommendations.

Sterilization: validated process used to render a product free of all forms of viable microorganisms. In a sterilization process, the presence of microorganisms on any individual item can be expressed in terms of probability. Although this probability can be reduced to a very low number, it can never be reduced to zero.

Sterilization area: area of a health-care facility designed to house sterilization equipment, such as steam ethylene oxide, hydrogen peroxide gas plasma, or ozone sterilizers.

Sterilizer: apparatus used to sterilize medical devices, equipment, or supplies by direct exposure to the sterilizing agent.

Sterilizer, gravity-displacement type: type of steam sterilizer in which incoming steam displaces residual air through a port or drain in or near the bottom (usually) of the sterilizer chamber. Typical operating temperatures are 121–123°C (250–254°F) and 132–135°C (270–275°F).

Sterilizer, prevacuum type: type of steam sterilizer that depends on one or more pressure and vacuum excursions at the beginning of the cycle to remove air. This method of operation results in shorter cycle times for wrapped items because of the rapid removal of air from the chamber and the load by the vacuum system and because of the usually higher operating temperature (132–135°C [270–275°F]; 141–144°C [285–291°F]). This type of sterilizer generally provides for shorter exposure time and accelerated drying of fabric loads by pulling a further vacuum at the end of the sterilizing cycle.

Sterilizer, steam-flush pressure-pulse type: type of sterilizer in which a repeated sequence consisting of a steam flush and a pressure pulse removes air from the sterilizing chamber and processed materials using steam at above atmospheric pressure (no vacuum is required). Like a prevacuum sterilizer, a steam-flush pressure-pulse sterilizer rapidly removes air from the sterilizing chamber and wrapped items; however, the system is not susceptible to air leaks because air is removed with the sterilizing chamber pressure at above atmospheric pressure. Typical operating temperatures are 121–123°C (250–254°F), 132–135°C (270–275°F), and 141–144°C (285–291°F).

Surfactant: agent that reduces the surface tension of water or the tension at the interface between water and another liquid; a wetting agent found in many sterilants and disinfectants.

Tabletop steam sterilizer: a compact gravity-displacement steam sterilizer that has a chamber volume of not more than 2 cubic feet and that generates its own steam when distilled or deionized water is added.

Time-weighted average (TWA): an average of all the concentrations of a chemical to which a worker has been exposed during a specific sampling time, reported as an average over the sampling time. For example, the permissible exposure limit for ethylene oxide is 1 ppm as an 8-hour TWA. Exposures above the ppm limit are permitted if they are compensated for by equal or longer exposures below the limit during the 8-hour workday as long as they do not exceed the ceiling limit; short-term exposure limit; or, in the case of ethylene oxide, excursion limit of 5 ppm averaged over a 15-minute sampling period.

Tuberculocide: an EPA-classified hospital disinfectant that also kills *Mycobacterium tuberculosis* (tubercle bacilli). EPA has registered approximately 200 tuberculocides. Such agents also are called *mycobactericides*.

Use-life: the length of time a diluted product can remain active and effective. The stability of the chemical and the storage conditions (e.g., temperature and presence of air, light, organic matter, or metals)

determine the use-life of antimicrobial products.

Vegetative bacteria: bacteria that are devoid of spores and usually can be readily inactivated by many types of germicides.

Virucide: an agent that kills viruses to make them noninfective.

Adapted from Association for the Advancement of Medical Instrumentation; [811-814, 819] Association of periOperating Registered Nurses (AORN), [815] American Hospital Association, [319] and Block. [16, 1034]

Table 1. Methods of sterilization and disinfection.

	Sterilization		Disinfection		
	Critical items (will enter tissue or vascular system or blood will flow through them)		High-level (semicritical items; [except dental] will come in contact with mucous membrane or nonintact skin)	Intermediate-level (some semicritical items[1] and noncritical items)	Low-level (noncritical items; will come in contact with intact skin)
Object	**Procedure**	**Exposure time**	**Procedure (exposure time 12-30 min at ≥20°C)[2,3]**	**Procedure (exposure time ≥ 1 m)[9]**	**Procedure (exposure time ≥ 1 m)[9]**
Smooth, hard Surface[1,4]	A	MR	D	K	K
	B	MR	E	L[5]	L
	C	MR	F	M	M
	D	10 h at 20-25°C	H	N	N
	F	6 h	I[6]		O
	G	12 m at 50-56°C	J		
	H	3-8 h			
Rubber tubing and catheters[3,4]	A	MR	D		
	B	MR	E		
	C	MR	F		
	D	10 h at 20-25°C	H		
	F	6 h	I[6]		
	G	12 m at 50-56°C	J		
	H	3-8 h			
Polyethylene tubing and catheters[3,4,7]	A	MR	D		
	B	MR	E		
	C	MR	F		
	D	10 h at 20-25°C	H		
	F	6 h	I[6]		
	G	12 m at 50-56°C	J		
	H	3-8 h			
Lensed instruments[4]	A	MR	D		
	B	MR	E		
	C	MR	F		
	D	10 h at 20-25°C	H		
	F	6 h	J		
	G	12 m at 50-56°C			
	H	3-8 h			
Thermometers (oral and rectal)[8]					K[8]
Hinged instruments[4]	A	MR	D		
	B	MR	E		
	C	MR	F		
	D	10 h at 20-25°C	H		
	F	6 h	I[6]		
	G	12 m at 50-56°C	J		
	H	3-8 h			

Modified from Rutala and Simmons. [15, 17, 18, 421] The selection and use of disinfectants in the healthcare field is dynamic, and products may become available that are not in existence when this guideline was written. As newer disinfectants become available, persons or committees responsible for selecting disinfectants and sterilization processes should be guided by products cleared by the FDA and the EPA as well as information in the scientific literature.

A, Heat sterilization, including steam or hot air (see manufacturer's recommendations, steam sterilization processing time from 3-30 minutes)
B, Ethylene oxide gas (see manufacturer's recommendations, generally 1-6 hours processing time plus aeration time of 8-12 hours at 50-60°C)
C, Hydrogen peroxide gas plasma (see manufacturer's recommendations for internal diameter and length restrictions, processing time between 45-72 minutes).
D, Glutaraldehyde-based formulations (≥2% glutaraldehyde, caution should be exercised with all glutaraldehyde formulations when further in-use dilution is anticipated); glutaraldehyde (1.12%) and 1.93% phenol/phenate. One glutaraldehyde-based product has a high-level disinfection claim of 5 minutes at 35°C.
E, Ortho-phthalaldehyde (OPA) 0.55%
F, Hydrogen peroxide 7.5% (will corrode copper, zinc, and brass)
G, Peracetic acid, concentration variable but 0.2% or greater is sporicidal. Peracetic acid immersion system operates at 50-56°C.
H, Hydrogen peroxide (7.35%) and 0.23% peracetic acid; hydrogen peroxide 1% and peracetic acid 0.08% (will corrode metal instruments)
I, Wet pasteurization at 70°C for 30 minutes with detergent cleaning
J, Hypochlorite, single use chlorine generated on-site by electrolyzing saline containing >650-675 active free chlorine; (will corrode metal instruments)
K, Ethyl or isopropyl alcohol (70-90%)
L, Sodium hypochlorite (5.25-6.15% household bleach diluted 1:500 provides **>**100 ppm available chlorine)
M, Phenolic germicidal detergent solution (follow product label for use-dilution)
N, Iodophor germicidal detergent solution (follow product label for use-dilution)
O, Quaternary ammonium germicidal detergent solution (follow product label for use-dilution)
MR, Manufacturer's recommendations
NA, Not applicable

[1] See text for discussion of hydrotherapy.
[2] The longer the exposure to a disinfectant, the more likely it is that all microorganisms will be eliminated. Follow the FDA-cleared high-level disinfection claim. Ten-minute exposure is not adequate to disinfect many objects, especially those that are difficult to clean because they have narrow channels or other areas that can harbor organic material and bacteria. Twenty-minute exposure at 20°C is the minimum time needed to reliably kill *M. tuberculosis* and nontuberculous mycobacteria with a 2% glutaraldehyde. Some high-level disinfectants have a reduced exposure time (e.g., ortho-phthalaldehyde at 12 minutes at 20°C) because of their rapid activity against mycobacteria or reduced exposure time due to increased mycobactericidal activity at elevated temperature (e.g., 2.5% glutaraldehyde at 5 minutes at 35°C, 0.55% OPA at 5 min at 25°C in automated endoscope reprocessor).
[3] Tubing must be completely filled for high-level disinfection and liquid chemical sterilization; care must be taken to avoid entrapment of air bubbles during immersion.
[4] Material compatibility should be investigated when appropriate.
[5] A concentration of 1000 ppm available chlorine should be considered where cultures or concentrated preparations of microorganisms have spilled (5.25% to 6.15% household bleach diluted 1:50 provides > 1000 ppm available chlorine). This solution may corrode some surfaces.
[6] Pasteurization (washer-disinfector) of respiratory therapy or anesthesia equipment is a recognized alternative to high-level disinfection. Some data challenge the efficacy of some pasteurization units.
[7] Thermostability should be investigated when appropriate.
[8] Do not mix rectal and oral thermometers at any stage of handling or processing.
[9] By law, all applicable label instructions on EPA-registered products must be followed. If the user selects exposure conditions that differ from those on the EPA-registered products label, the user assumes liability from any injuries resulting from off-label use and is potentially subject to enforcement action under FIFRA.

Table 2. Properties of an ideal disinfectant.

Broad spectrum: should have a wide antimicrobial spectrum
Fast acting: should produce a rapid kill
Not affected by environmental factors: should be active in the presence of organic matter (e.g., blood, sputum, feces) and compatible with soaps, detergents, and other chemicals encountered in use
Nontoxic: should not be harmful to the user or patient
Surface compatibility: should not corrode instruments and metallic surfaces and should not cause the deterioration of cloth, rubber, plastics, and other materials
Residual effect on treated surfaces: should leave an antimicrobial film on the treated surface
Easy to use with clear label directions
Odorless: should have a pleasant odor or no odor to facilitate its routine use
Economical: should not be prohibitively high in cost
Solubility: should be soluble in water
Stability: should be stable in concentrate and use-dilution
Cleaner: should have good cleaning properties
Environmentally friendly: should not damage the environment on disposal

Modified from Molinari[1035].

Table 3. Epidemiologic evidence associated with the use of surface disinfectants or detergents on noncritical environmental surfaces.

Justification for Use of Disinfectants for Noncritical Environmental Surfaces

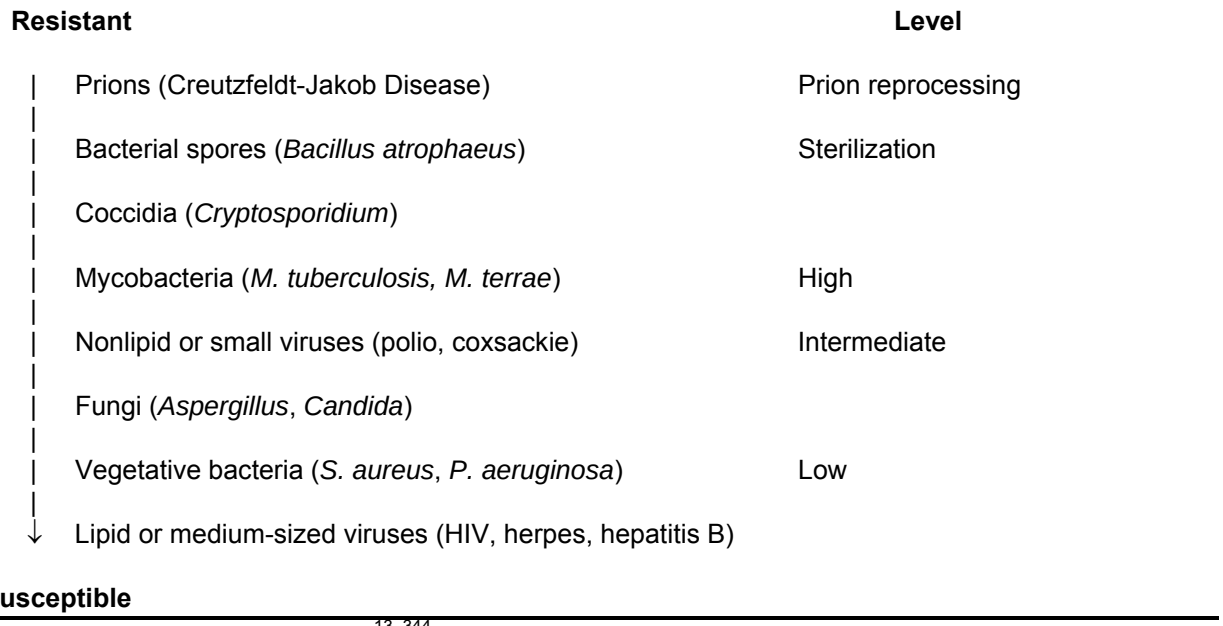
Surfaces may contribute to transmission of epidemiologically important microbes (e.g., vancomycin-resistant Enterococci, methicillin-resistant *S. aureus*, viruses)
Disinfectants are needed for surfaces contaminated by blood and other potentially infective material
Disinfectants are more effective than detergents in reducing microbial load on floors
Detergents become contaminated and result in seeding the patient's environment with bacteria
Disinfection of noncritical equipment and surfaces is recommended for patients on isolation precautions by the Centers for Disease Control and Prevention.
Advantage of using a single product for decontamination of noncritical surfaces, both floors and equipment
Some newer disinfectants have persistent antimicrobial activity

Justification for Using a Detergent on Noncritical Environmental Surfaces

Noncritical surfaces contribute minimally to endemic healthcare-associated infections
No difference in healthcare-associated infection rates when floors are cleaned with detergent versus disinfectant
No environmental impact (aquatic or terrestrial) issues with disposal
No occupational health exposure issues
Lower costs
Use of antiseptics/disinfectants selects for antibiotic-resistant bacteria (?)
More aesthetically pleasing floor

Modified from Rutala[378].

Figure 1. Decreasing order of resistance of microorganisms to disinfection and sterilization and the level of disinfection or sterilization.



Modified from Russell and Favero [13, 344].

Table 4. Comparison of the characteristics of selected chemicals used as high-level disinfectants or chemical sterilants.

	HP (7.5%)	PA (0.2%)	Glut (≥2.0%)	OPA (0.55%)	HP/PA (7.35%/0.23%)
HLD Claim	30 m @ 20°C	NA	20-90 m @ 20°-25°C	12 m @ 20°C, 5 m @ 25°C in AER	15m @ 20°C
Sterilization Claim	6 h @ 20°	12m @ 50-56°C	10 h @ 20°-25°C	None	3 h @ 20°C
Activation	No	No	Yes (alkaline glut)	No	No
Reuse Life[1]	21d	Single use	14-30 d	14d	14d
Shelf Life Stability[2]	2 y	6 mo	2 y	2 y	2 y
Disposal Restrictions	None	None	Local[3]	Local[3]	None
Materials Compatibility	Good	Good	Excellent	Excellent	No data
Monitor MEC[4]	Yes (6%)	No	Yes (1.5% or higher)	Yes (0.3% OPA)	No
Safety	Serious eye damage (safety glasses)	Serious eye and skin damage (conc soln)[5]	Respiratory	Eye irritant, stains skin	Eye damage
Processing	Manual or automated	Automated	Manual or automated	Manual or automated	Manual
Organic material resistance	Yes	Yes	Yes	Yes	Yes
OSHA exposure limit	1 ppm TWA	None	None[6]	None	HP-1 ppm TWA
Cost profile (per cycle)[7]	+ (manual), ++ (automated)	+++++ (automated)	+ (manual), ++ (automated)	++ (manual)	++ (manual)

Modified from Rutala [69].

Abbreviations: HLD=high-level disinfectant; HP=hydrogen peroxide; PA=peracetic acid; glut=glutaraldehyde; PA/HP=peracetic acid and hydrogen peroxide; OPA =ortho-phthalaldehyde (FDA cleared as a high-level disinfectant, included for comparison to other chemical agents used for high-level disinfection); m=minutes; h=hours; NA=not applicable; TWA=time-weighted average for a conventional 8-hour workday.

[1]number of days a product can be reused as determined by re-use protocol

[2]time a product can remain in storage (unused)

[3]no U.S. EPA regulations but some states and local authorities have additional restrictions

[4]MEC=minimum effective concentration is the lowest concentration of active ingredients at which the product is still effective

[5]Conc soln=concentrated solution

[6]The ceiling limit recommended by the American Conference of Governmental Industrial Hygienists is 0.05 ppm.

[7]per cycle cost profile considers cost of the processing solution (suggested list price to healthcare facilities in August 2001) and assumes maximum use life (e.g., 21 days for hydrogen peroxide, 14 days for glutaraldehyde), 5 reprocessing cycles per day, 1-gallon basin for manual processing, and 4-gallon tank for automated processing. + = least expensive; +++++ = most expensive

Table 5. Summary of advantages and disadvantages of chemical agents used as chemical sterilants[1] or as high-level disinfectants.		
Sterilization Method	Advantages	Disadvantages
Peracetic Acid/Hydrogen Peroxide	• No activation required • Odor or irritation not significant	• Materials compatibility concerns (lead, brass, copper, zinc) both cosmetic and functional • Limited clinical experience • Potential for eye and skin damage
Glutaraldehyde	• Numerous use studies published • Relatively inexpensive • Excellent materials compatibility	• Respiratory irritation from glutaraldehyde vapor • Pungent and irritating odor • Relatively slow mycobactericidal activity • Coagulates blood and fixes tissue to surfaces • Allergic contact dermatitis • Glutaraldehyde vapor monitoring recommended
Hydrogen Peroxide	• No activation required • May enhance removal of organic matter and organisms • No disposal issues • No odor or irritation issues • Does not coagulate blood or fix tissues to surfaces • Inactivates *Cryptosporidium* • Use studies published	• Material compatibility concerns (brass, zinc, copper, and nickel/silver plating) both cosmetic and functional • Serious eye damage with contact
Ortho-phthalaldehyde	• Fast acting high-level disinfectant • No activation required • Odor not significant • Excellent materials compatibility claimed • Does not coagulate blood or fix tissues to surfaces claimed	• Stains skin, mucous membranes, clothing, and environmental surfaces • Repeated exposure may result in hypersensitivity in some patients with bladder cancer • More expensive than glutaraldehyde • Eye irritation with contact • Slow sporicidal activity
Peracetic Acid	• Rapid sterilization cycle time (30-45 minutes) • Low temperature (50-55°C) liquid immersion sterilization • Environmental friendly by-products (acetic acid, O_2, H_20) • Fully automated • Single-use system eliminates need for concentration testing • Standardized cycle • May enhance removal of organic material and endotoxin • No adverse health effects to operators under normal operating conditions • Compatible with many materials and instruments • Does not coagulate blood or fix tissues to surfaces • Sterilant flows through scope facilitating salt, protein, and microbe removal • Rapidly sporicidal • Provides procedure standardization (constant dilution, perfusion of channel, temperatures, exposure)	• Potential material incompatibility (e.g., aluminum anodized coating becomes dull) • Used for immersible instruments only • Biological indicator may not be suitable for routine monitoring • One scope or a small number of instruments can be processed in a cycle • More expensive (endoscope repairs, operating costs, purchase costs) than high-level disinfection • Serious eye and skin damage (concentrated solution) with contact • Point-of-use system, no sterile storage

Modified from Rutala[69].

[1]All products effective in presence of organic soil, relatively easy to use, and have a broad spectrum of antimicrobial activity (bacteria, fungi, viruses, bacterial spores, and mycobacteria). The above characteristics are documented in the literature; contact the manufacturer of the instrument and sterilant for additional information. All products listed above are FDA-cleared as chemical sterilants except OPA, which is an FDA-cleared high-level disinfectant.

Table 6. Summary of advantages and disadvantages of commonly used sterilization technologies.

Sterilization Method	Advantages	Disadvantages
Steam	· Nontoxic to patient, staff, environment · Cycle easy to control and monitor · Rapidly microbicidal · Least affected by organic/inorganic soils among sterilization processes listed · Rapid cycle time · Penetrates medical packing, device lumens	· Deleterious for heat-sensitive instruments · Microsurgical instruments damaged by repeated exposure · May leave instruments wet, causing them to rust • Potential for burns
Hydrogen Peroxide Gas Plasma	· Safe for the environment · Leaves no toxic residuals · Cycle time is 28-75 minutes (varies with model type) and no aeration necessary · Used for heat- and moisture-sensitive items since process temperature <50°C · Simple to operate, install (208 V outlet), and monitor · Compatible with most medical devices . Only requires electrical outlet	· Cellulose (paper), linens and liquids cannot be processed · Sterilization chamber size from 1.8-9.4 ft^3 total volume (varies with model type) · Some endoscopes or medical devices with long or narrow lumens cannot be processed at this time in the United States (see manufacturer's recommendations for internal diameter and length restrictions) · Requires synthetic packaging (polypropylene wraps, polyolefin pouches) and special container tray • Hydrogen peroxide may be toxic at levels greater than 1 ppmTWA
100% Ethylene Oxide (ETO)	· Penetrates packaging materials, device lumens · Single-dose cartridge and negative- pressure chamber minimizes the potential for gas leak and ETO exposure · Simple to operate and monitor · Compatible with most medical materials	· Requires aeration time to remove ETO residue · Sterilization chamber size from 4.0-7.9 ft^3 total volume (varies with model type) · ETO is toxic, a carcinogen, and flammable · ETO emission regulated by states but catalytic cell removes 99.9% of ETO and converts it to CO_2 and H_2O · ETO cartridges should be stored in flammable liquid storage cabinet · Lengthy cycle/aeration time
ETO Mixtures 8.6% ETO/91.4% HCFC 10% ETO/90% HCFC 8.5% ETO/91.5% CO_2	· Penetrates medical packaging and many plastics · Compatible with most medical materials · Cycle easy to control and monitor	· Some states (e.g., CA, NY, MI) require ETO emission reduction of 90-99.9% · CFC (inert gas that eliminates explosion hazard) banned in 1995 · Potential hazards to staff and patients · Lengthy cycle/aeration time . ETO is toxic, a carcinogen, and flammable
Peracetic Acid	· Rapid cycle time (30-45 minutes) Low temperature (50-55°C liquid immersion sterilization · Environmental friendly by-products · Sterilant flows through endoscope which facilitates salt, protein and microbe removal	· Point-of-use system, no sterile storage · Biological indicator may not be suitable for routine monitoring · Used for immersible instruments only · Some material incompatibility (e.g., aluminum anodized coating becomes dull) · One scope or a small number of instruments processed in a cycle • Potential for serious eye and skin damage (concentrated solution) with contact

Modified from Rutala. [825]

Abbreviations: CFC=chlorofluorocarbon, HCFC=hydrochlorofluorocarbon.

Table 7. Minimum cycle times for steam sterilization cycles

Type of sterilizer	Item	Exposure time at 250°F (121°C)	Exposure time at 270°F (132°C)	Drying time
Gravity displacement	Wrapped instruments	30 min	15 min	**15-30** min
	Textile packs	30 min	25 min	**15** min
	Wrapped utensils	30 min	15 min	**15-30** min
Dynamic-air-removal (e.g., prevacuum)	Wrapped instruments		4 min	**20-30** min
	Textile packs		4 min	**5-20** min
	Wrapped utensils		4 min	20 min

Modified from Association for the Advancement of Medical Instrumentation. [813, 819]

Table 8. Examples of flash steam sterilization parameters.

Type of sterilizer	Load configuration	Temperature	Time
Gravity displacement	Nonporous items only (i.e., routine metal instruments, no lumens)	132°C (270°F)	3 minutes
	Nonporous and porous items (e.g., rubber or plastic items, items with lumens) sterilized together	132°C (270°F)	10 minutes
Prevacuum	Nonporous items only (i.e., routine metal instruments, no lumens)	132°C (270°F)	3 minutes
	Nonporous and porous items (e.g., rubber or plastic items, items with lumens) sterilized together	132°C (270°F)	4 minutes
Steam-flush pressure-pulse	Nonporous or mixed nonporous/porous items	132° (270°F) Manufacturers' instruction	**4** minutes

Modified from Association for the Advancement of Medical Instrumentation. [812, 819]

Table 9. Characteristics of an ideal low-temperature sterilization process.

High efficacy: the agent should be virucidal, bactericidal, tuberculocidal, fungicidal and sporicidal
Rapid activity: ability to quickly achieve sterilization
Strong penetrability: ability to penetrate common medical-device packaging materials and penetrate into the interior of device lumens
Material compatibility: produces only negligible changes in the appearance or the function of processed items and packaging materials even after repeated cycling
Nontoxic: presents no toxic health risk to the operator or the patient and poses no hazard to the environment
Organic material resistance: withstands reasonable organic material challenge without loss of efficacy
Adaptability: suitable for large or small (point of use) installations
Monitoring capability: monitored easily and accurately with physical, chemical, and biological process monitors
Cost effectiveness: reasonable cost for installation and for routine operation

Modified from Schneider. [851]

Table 10. Factors affecting the efficacy of sterilization.

Factors	Effect
Cleaning[1]	Failure to adequately clean instrument results in higher bioburden, protein load, and salt concentration. These will decrease sterilization efficacy.
Bioburden[1]	The natural bioburden of used surgical devices is 10^0 to 10^3 organisms (primarily vegetative bacteria), which is substantially below the 10^5-10^6 spores used with biological indicators.
Pathogen type	Spore-forming organisms are most resistant to sterilization and are the test organisms required for FDA clearance. However, the contaminating microflora on used surgical instruments consists mainly of vegetative bacteria.
Protein[1]	Residual protein decreases efficacy of sterilization. However, cleaning appears to rapidly remove protein load.
Salt[1]	Residual salt decreases efficacy of sterilization more than does protein load. However, cleaning appears to rapidly remove salt load.
Biofilm accumulation[1]	Biofilm accumulation reduces efficacy of sterilization by impairing exposure of the sterilant to the microbial cell.
Lumen length	Increasing lumen length impairs sterilant penetration. May require forced flow through lumen to achieve sterilization.
Lumen diameter	Decreasing lumen diameter impairs sterilant penetration. May require forced flow through lumen to achieve sterilization.
Restricted flow	Sterilant must come into contact with microorganisms. Device designs that prevent or inhibit this contact (e.g., sharp bends, blind lumens) will decrease sterilization efficacy.
Device design and construction	Materials used in construction may affect compatibility with different sterilization processes and affect sterilization efficacy. Design issues (e.g., screws, hinges) will also affect sterilization efficacy.

Modified from Alfa and Rutala. [470, 825] [1] Factor only relevant for reused surgical/medical devices

Table 11. Comparative evaluation of the microbicidal activity of low-temperature sterilization technology.

Challenge	Carriers Sterilized by Various Low-Temperature Sterilization Technologies						
	ETO 12/88	100% ETO	HCFC-ETO	HPGP 100	HPGP 100S	PA	Reference
No salt or serum[1]	100%	100%	96%	100%	ND	ND	Alfa [721]
10% serum and 0.65% salt[2]	97%	60%	95%	37%	ND	ND	Alfa [721]
Lumen (125 cm long x 3 mm wide) without serum or salt[1]	ND	96%	96%	ND	ND	ND	Alfa [721]
Lumen (125 cm long x 3 mm wide) with 10% serum and 0.65% salt[2]	44%	40%	49%	35%	ND	100%[1]	Alfa [721]
Lumen (40 cm long x 3 mm wide)[3]	ND	ND	100%	95%	100%	8%	Rutala [856]
Lumen (40 cm long x 2 mm wide)[3]	ND	ND	100%	93%	100%	ND	Rutala [856]
Lumen (40 cm long x 1 mm wide)[3]	ND	ND	100%	26%	100%	ND	Rutala [856]
Lumen (40 cm long x 3 mm wide)[4]	ND	ND	100%	100%	100%	ND	Rutala [856]

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Modified from Rutala. [825]
Abbreviations: ETO=ethylene oxide; HCFC=hydrochlorofluorocarbon; ND=no data; HPGP=hydrogen peroxide gas plasma; PA=peracetic acid.

[1]Test organisms included *Enterococcus faecalis*, *Mycobacterium chelonae*, and *Bacillus atrophaeus* spores.
[2]Test organisms included *E. faecalis*, *P. aeruginosa*, *E. coli*, *M. chelonae*, *B. atrophaeus* spores, *G. stearothermophilus* spores, and *B. circulans* spores.
[3]Test organism was *G. stearothermophilus* spores . The lumen test units had a removable 5 cm center piece (1.2 cm diameter) of stainless steel sealed to the narrower steel tubing by hard rubber septums.
[4]Test organism was *G. stearothermophilus* spores. The lumen test unit was a straight stainless steel tube.

Table 12. Suggested protocol for management of positive biological indicator in a steam sterilizer.

1. Take the sterilizer out of service. Notify area supervisor and infection control department.
2. Objects, other than implantable objects, do not need to be recalled because of a single positive spore test unless the sterilizer or the sterilization procedure is defective. As soon as possible, repeat biological indicator test in three consecutive sterilizer cycles. If additional spore tests remain positive, the items should be considered nonsterile, and supplies processed since the last acceptable (negative) biological indicator should be recalled. The items from the suspect load(s) should be recalled and reprocessed.
3. Check to ensure the sterilizer was used correctly (e.g., verify correct time and temperature setting). If not, repeat using appropriate settings and recall and reprocess all inadequately processed items.
4. Check with hospital maintenance for irregularities (e.g., electrical) or changes in the hospital steam supply (i.e., from standard $\geq$97% steam, <3% moisture). Any abnormalities should be reported to the person who performs sterilizer maintenance (e.g., medical engineering, sterilizer manufacturer).
5. Check to ensure the correct biological indicator was used and appropriately interpreted. If not, repeat using appropriate settings.

If steps 1 through 5 resolve the problem

6. If all three repeat biological indicators from three consecutive sterilizer cycles (step 2 above) are negative, put the sterilizer back in service.

If one or both biological indicators are positive, do one or more of the following until problem is resolved.

7. A. Request an inspection of the equipment by sterilizer maintenance personnel.
   B. Have hospital maintenance inspect the steam supply lines.
   C. Discuss the abnormalities with the sterilizer manufacturer.
   D. Repeat the biological indicator using a different manufacturer's indicator.

If step 7 does not resolve the problem

Close sterilizer down until the manufacturer can assure that it is operating properly. Retest at that time with biological indicators in three consecutive sterilizer cycles.

Modified from Bryce. [839]

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2007 Guideline for Isolation Precautions: Preventing Transmission of Infectious Agents in Healthcare Settings

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EXECUTIVE SUMMARY

The *Guideline for Isolation Precautions: Preventing Transmission of Infectious Agents in Healthcare Settings 2007* updates and expands the *1996 Guideline for Isolation Precautions in Hospitals*. The following developments led to revision of the 1996 guideline:

1. The transition of healthcare delivery from primarily acute care hospitals to other healthcare settings (e.g., home care, ambulatory care, free-standing specialty care sites, long-term care) created a need for recommendations that can be applied in all healthcare settings using common principles of infection control practice, yet can be modified to reflect setting-specific needs. Accordingly, the revised guideline addresses the spectrum of healthcare delivery settings. Furthermore, the term “nosocomial infections“ is replaced by “healthcare-associated infections” (HAIs) to reflect the changing patterns in healthcare delivery and difficulty in determining the geographic site of exposure to an infectious agent and/or acquisition of infection.
2. The emergence of new pathogens (e.g., SARS-CoV associated with the severe acute respiratory syndrome [SARS], Avian influenza in humans), renewed concern for evolving known pathogens (e.g., *C. difficile,* noroviruses, community-associated MRSA [CA-MRSA]), development of new therapies (e.g., gene therapy), and increasing concern for the threat of bioweapons attacks, established a need to address a broader scope of issues than in previous isolation guidelines.
3. The successful experience with Standard Precautions, first recommended in the 1996 guideline, has led to a reaffirmation of this approach as the foundation for preventing transmission of infectious agents in all healthcare settings. New additions to the recommendations for Standard Precautions are Respiratory Hygiene/Cough Etiquette and safe injection practices, including the use of a mask when performing certain high-risk, prolonged procedures involving spinal canal punctures (e.g., myelography, epidural anesthesia). The need for a recommendation for Respiratory Hygiene/Cough Etiquette grew out of observations during the SARS outbreaks where failure to implement simple source control measures with patients, visitors, and healthcare personnel with respiratory symptoms may have contributed to SARS coronavirus (SARS-CoV) transmission. The recommended practices have a strong evidence base. The continued occurrence of outbreaks of hepatitis B and hepatitis C viruses in ambulatory settings indicated a need to re-iterate safe injection practice recommendations as part of Standard Precautions. The addition of a mask for certain spinal injections grew from recent evidence of an associated risk for developing meningitis caused by respiratory flora.
4. The accumulated evidence that environmental controls decrease the risk of life-threatening fungal infections in the most severely immunocompromised patients (allogeneic hematopoietic stem-cell transplant patients) led to the update on the components of the Protective Environment (PE).
5. Evidence that organizational characteristics (e.g., nurse staffing levels and composition, establishment of a safety culture) influence healthcare personnel adherence to recommended infection control practices, and therefore are important factors in preventing transmission of infectious agents, led to a new

emphasis and recommendations for administrative involvement in the development and support of infection control programs.

6. Continued increase in the incidence of HAIs caused by multidrug-resistant organisms (MDROs) in all healthcare settings and the expanded body of knowledge concerning prevention of transmission of MDROs created a need for more specific recommendations for surveillance and control of these pathogens that would be practical and effective in various types of healthcare settings.

This document is intended for use by infection control staff, healthcare epidemiologists, healthcare administrators, nurses, other healthcare providers, and persons responsible for developing, implementing, and evaluating infection control programs for healthcare settings across the continuum of care. The reader is referred to other guidelines and websites for more detailed information and for recommendations concerning specialized infection control problems.

Parts I - III: Review of the Scientific Data Regarding Transmission of Infectious Agents in Healthcare Settings Part I reviews the relevant scientific literature that supports the recommended prevention and control practices. As with the 1996 guideline, the modes and factors that influence transmission risks are described in detail. New to the section on transmission are discussions of bioaerosols and of how droplet and airborne transmission may contribute to infection transmission. This became a concern during the SARS outbreaks of 2003, when transmission associated with aerosol-generating procedures was observed. Also new is a definition of "epidemiologically important organisms" that was developed to assist in the identification of clusters of infections that require investigation (i.e. multidrug-resistant organisms, *C. difficile)*. Several other pathogens that hold special infection control interest (i.e., norovirus, SARS, Category A bioterrorist agents, prions, monkeypox, and the hemorrhagic fever viruses) also are discussed to present new information and infection control lessons learned from experience with these agents. This section of the guideline also presents information on infection risks associated with specific healthcare settings and patient populations.

Part II updates information on the basic principles of hand hygiene, barrier precautions, safe work practices and isolation practices that were included in previous guidelines. However, new to this guideline, is important information on healthcare system components that influence transmission risks, including those under the influence of healthcare administrators. An important administrative priority that is described is the need for appropriate infection control staffing to meet the ever-expanding role of infection control professionals in the modern, complex healthcare system. Evidence presented also demonstrates another administrative concern, the importance of nurse staffing levels, including numbers of appropriately trained nurses in ICUs for preventing HAIs. The role of the clinical microbiology laboratory in supporting infection control is described to emphasize the need for this service in healthcare facilites. Other factors that influence transmission risks are discussed i.e., healthcare worker adherence to recommended infection control practices, organizational safety culture or climate, education and training
Discussed for the first time in an isolation guideline is surveillance of healthcare-associated infections. The information presented will be useful to new infection control professionals as

well as persons involved in designing or responding to state programs for public reporting of HAI rates.

Part III describes each of the categories of precautions developed by the Healthcare Infection Control Practices Advisory Committee (HICPAC) and the Centers for Disease Control and Prevention (CDC) and provides guidance for their application in various healthcare settings. The categories of Transmission-Based Precautions are unchanged from those in the 1996 guideline: Contact, Droplet, and Airborne. One important change is the recommendation to don the indicated personal protective equipment (gowns, gloves, mask) *upon entry into the patient's room* for patients who are on Contact and/or Droplet Precautions since the nature of the interaction with the patient cannot be predicted with certainty and contaminated environmental surfaces are important sources for transmission of pathogens.
In addition, the Protective Environment (PE) for allogeneic hematopoietic stem cell transplant patients, described in previous guidelines, has been updated.

Tables, Appendices, and other Information
There are several tables that summarize important information: 1) a summary of the evolution of this document; 2) guidance on using empiric isolation precautions according to a clinical syndrome; 3) a summary of infection control recommendations for category A agents of bioterrorism; 4) components of Standard Precautions and recommendations for their application; 5) components of the Protective Environment; and 6) a glossary of definitions used in this guideline. New in this guideline is a figure that shows a recommended sequence for donning and removing personal protective equipment used for isolation precautions to optimize safety and prevent self-contamination during removal.

Appendix A: Type and Duration of Precautions Recommended for Selected Infections and Conditions
Appendix A consists of an updated alphabetical list of most infectious agents and clinical conditions for which isolation precautions are recommended. A preamble to the Appendix provides a rationale for recommending the use of one or more Transmission-Based Precautions, in addition to Standard Precautions, based on a review of the literature and evidence demonstrating a real or potential risk for person-to-person transmission in healthcare settings.The type and duration of recommended precautions are presented with additional comments concerning the use of adjunctive measures or other relevant considerations to prevent transmission of the specific agent. Relevant citations are included.

Pre- Publication of the Guideline on Preventing Transmission of MDROs
New to this guideline is a comprehensive review and detailed recommendations for prevention of transmission of MDROs. This portion of the guideline was published electronically in October 2006 and updated in November, 2006 (Siegel JD, Rhinehart E, Jackson M, Chiarello L and HICPAC. Management of Multidrug-Resistant Organisms in Healthcare Settings 2006 www.cdc.gov/ncidod/dhqp/pdf/ar/mdroGuideline2006.pdf), and is considered a part of the Guideline for Isolation Precautions. This section provides a detailed review of the complex topic of MDRO control in healthcare settings and is intended to provide a context for evaluation of MDRO at individual healthcare settings. A rationale and

institutional requirements for developing an effective MDRO control program are summarized. Although the focus of this guideline is on measures to prevent *transmission* of MDROs in healthcare settings, information concerning the judicious use of antimicrobial agents is presented since such practices are intricately related to the size of the reservoir of MDROs which in turn influences transmission (e.g. colonization pressure). There are two tables that summarize recommended prevention and control practices using the following seven categories of interventions to control MDROs: administrative measures, education of healthcare personnel, judicious antimicrobial use, surveillance, infection control precautions, environmental measures, and decolonization. Recommendations for each category apply to and are adapted for the various healthcare settings. With the increasing incidence and prevalence of MDROs, all healthcare facilities must prioritize effective control of MDRO transmission. Facilities should identify prevalent MDROs at the facility, implement control measures, assess the effectiveness of control programs, and demonstrate decreasing MDRO rates. A set of intensified MDRO prevention interventions is presented to be added 1) if the incidence of transmission of a target MDRO is NOT decreasing despite implementation of basic MDRO infection control measures, and 2) when the *first* case(s) of an epidemiologically important MDRO is identified within a healthcare facility.

Summary

This updated guideline responds to changes in healthcare delivery and addresses new concerns about transmission of infectious agents to patients and healthcare workers in the United States and infection control. The primary objective of the guideline is to improve the safety of the nation's healthcare delivery system by reducing the rates of HAIs.

Abbreviations Used in the Guideline

AIIR	Airborne infection isolation room
CDC	Centers for Disease Control and Prevention
CF	Cystic fibrosis
CJD	Creutzfeld-Jakob Disease
CLSI	Clinical Laboratory Standards Institute
ESBL	Extended spectrum beta-lactamases
FDA	Food and Drug Administration
HAI	Healthcare-associated infections
HBV	Hepatitis B virus
HCV	Hepatitis C virus
HEPA	High efficiency particulate air [filtration]
HICPAC	Healthcare Infection Control Practices Advisory Committee
HIV	Human immunodeficiency virus
HCW	Healthcare worker
HSCT	Hematopoetic stem-cell transplant
ICU	Intensive care unit
LTCF	Long-term care facility
MDRO	Multidrug-resistant organism
MDR-GNB	Multidrug-resistant gram-negative bacilli
MRSA	Methicillin-resistant *Staphylococcus aureus*
NCCLS	National Committee for Clinical Laboratory Standards
NICU	Neonatal intensive care unit
NIOSH	National Institute for Occupational Safety and Health, CDC
NNIS	National Nosocomial Infection Surveillance
NSSP	Nonsusceptible *Streptococcus pneumoniae*
OSHA	Occupational Safety and Health Administration
PICU	Pediatric intensive care unit
PPE	Personal protective equipment
RSV	Respiratory syncytial virus
SARS	Severe acquired respiratory syndrome
vCJD	variant Creutzfeld-Jakob Disease
VRE	Vancomycin-resistant enterococci
WHO	World Health Organization

Part I:
Review of Scientific Data Regarding Transmission of Infectious Agents in Healthcare Settings

I.A. Evolution of the 2007 Document

The *Guideline for Isolation Precautions: Preventing Transmission of Infectious Agents in Healthcare Settings 2007* builds upon a series of isolation and infection prevention documents promulgated since 1970. These previous documents are summarized and referenced in Table 1 and in Part I of the *1996 Guideline for Isolation Precautions in Hospitals* [1].

Objectives and methods The objectives of this guideline are to 1) provide infection control recommendations for all components of the healthcare delivery system, including hospitals, long-term care facilities, ambulatory care, home care and hospice; 2) reaffirm Standard Precautions as the foundation for preventing transmission during patient care in all healthcare settings; 3) reaffirm the importance of implementing Transmission-Based Precautions based on the clinical presentation or syndrome and likely pathogens until the infectious etiology has been determined (Table 2); and 4) provide epidemiologically sound and, whenever possible, evidence-based recommendations.

This guideline is designed for use by individuals who are charged with administering infection control programs in hospitals and other healthcare settings. The information also will be useful for other healthcare personnel, healthcare administrators, and anyone needing information about infection control measures to prevent transmission of infectious agents. Commonly used abbreviations are provided on page 12 and terms used in the guideline are defined in the Glossary (page 137).

Med-line and Pub Med were used to search for relevant studies published in English, focusing on those published since 1996. Much of the evidence cited for preventing transmission of infectious agents in healthcare settings is derived from studies that used “quasi-experimental designs”, also referred to as nonrandomized, pre- post-intervention study designs [2]. Although these types of studies can provide valuable information regarding the effectiveness of various interventions, several factors decrease the certainty of attributing improved outcome to a specific intervention. These include: difficulties in controlling for important confounding variables; the use of multiple interventions during an outbreak; and results that are explained by the statistical principle of regression to the mean, (e.g., improvement over time without any intervention) [3].

Observational studies remain relevant and have been used to evaluate infection control interventions [4, 5]. The quality of studies, consistency of results and correlation with results from randomized, controlled trials when available were considered during the literature review and assignment of evidence-based categories (See Part IV: Recommendations) to the recommendations in this guideline. Several authors have summarized properties to consider when evaluating studies for the purpose of determining if the results should change practice or in designing new studies [2, 6, 7].

Changes or clarifications in terminology This guideline contains four changes in terminology from the 1996 guideline:

- The term *nosocomial infection* is retained to refer only to infections acquired in hospitals. The term *healthcare-associated infection* (HAI) is used to refer to infections associated with healthcare delivery in any setting (e.g., hospitals, long-term care facilities, ambulatory settings, home care). This term reflects the inability to determine with certainty where the pathogen is acquired since patients may be colonized with or exposed to potential pathogens outside of the healthcare setting, before receiving health care, or may develop infections caused by those pathogens when exposed to the conditions associated with delivery of healthcare. Additionally, patients frequently move among the various settings within a healthcare system [8].
- A new addition to the practice recommendations for Standard Precautions is *Respiratory Hygiene/Cough Etiquette*. While Standard Precautions generally apply to the recommended practices of healthcare personnel during patient care, Respiratory Hygiene/Cough Etiquette applies broadly to all persons who enter a healthcare setting, including healthcare personnel, patients and visitors. These recommendations evolved from observations during the SARS epidemic that failure to implement basic source control measures with patients, visitors, and healthcare personnel with signs and symptoms of respiratory tract infection may have contributed to SARS coronavirus (SARS-CoV) transmission. This concept has been incorporated into CDC planning documents for SARS and pandemic influenza [9, 10].
- The term *"Airborne Precautions"* has been supplemented with the term "*Airborne Infection Isolation Room (AIIR)*" for consistency with the *Guidelines for Environmental Infection Control in Healthcare Facilities* [11], the *Guidelines for Preventing the Transmission of Mycobacterium tuberculosis in Health-Care Settings 2005*[12] and the American Institute of Architects (AIA) guidelines for design and construction of hospitals, 2006 [13]
- A set of prevention measures termed *Protective Environment* has been added to the precautions used to prevent HAIs. These measures, which have been defined in other guidelines , consist of engineering and design interventions that decrease the risk of exposure to environmental fungi for severely immunocompromised allogeneic hematiopoietic stem cell transplant (HSCT) patients during their highest risk phase, usually the first 100 days post transplant, or longer in the presence of graft-versus-host disease [11, 13-15]. Recommendations for a Protective Environment apply only to acute care hospitals that provide care to HSCT patients.

Scope This guideline, like its predecessors, focuses primarily on interactions between patients and healthcare providers. The Guidelines for the Prevention of MDRO Infection were published separately in November 2006, and are available online at www.cdc.gov/ncidod/dhqp/index.html. Several other HICPAC

guidelines to prevent transmission of infectious agents associated with healthcare delivery are cited; e.g., *Guideline for Hand Hygiene*, *Guideline for Environmental Infection Control*, *Guideline for Prevention of Healthcare-Associated Pneumonia*, and *Guideline for Infection Control in Healthcare Personnel* [11, 14, 16, 17]. In combination, these provide comprehensive guidance on the primary infection control measures for ensuring a safe environment for patients and healthcare personnel.

This guideline does not discuss in detail specialized infection control issues in defined populations that are addressed elsewhere, (e.g., *Recommendations for Preventing Transmission of Infections among Chronic Hemodialysis Patients* , *Guidelines for Preventing the Transmission of Mycobacterium tuberculosis in Health-Care Facilities 2005*, *Guidelines for Infection Control in Dental Health-Care Settings and Infection Control Recommendations for Patients with Cystic Fibrosis* [12, 18-20]. An exception has been made by including abbreviated guidance for a Protective Environment used for allogeneic HSCT recipients because components of the Protective Environment have been more completely defined since publication of the *Guidelines for Preventing Opportunistic Infections Among HSCT Recipients in 2000* and the *Guideline for Environmental Infection Control in Healthcare Facilities* [11, 15].

I.B. Rationale for Standard and Transmission-Based Precautions in healthcare settings

Transmission of infectious agents within a healthcare setting requires three elements: a source (or reservoir) of infectious agents, a susceptible host with a portal of entry receptive to the agent, and a mode of transmission for the agent. This section describes the interrelationship of these elements in the epidemiology of HAIs.

I.B.1. Sources of infectious agents Infectious agents transmitted during healthcare derive primarily from human sources but inanimate environmental sources also are implicated in transmission. Human reservoirs include patients [20-28], healthcare personnel [29-35 17, 36-39], and household members and other visitors [40-45]. Such source individuals may have active infections, may be in the asymptomatic and/or incubation period of an infectious disease, or may be transiently or chronically colonized with pathogenic microorganisms, particularly in the respiratory and gastrointestinal tracts. The endogenous flora of patients (e.g., bacteria residing in the respiratory or gastrointestinal tract) also are the source of HAIs [46-54].

I.B.2. Susceptible hosts Infection is the result of a complex interrelationship between a potential host and an infectious agent. Most of the factors that influence infection and the occurrence and severity of disease are related to the host. However, characteristics of the host-agent interaction as it relates to

pathogenicity, virulence and antigenicity are also important, as are the infectious dose, mechanisms of disease production and route of exposure [55]. There is a spectrum of possible outcomes following exposure to an infectious agent. Some persons exposed to pathogenic microorganisms never develop symptomatic disease while others become severely ill and even die. Some individuals are prone to becoming transiently or permanently colonized but remain asymptomatic. Still others progress from colonization to symptomatic disease either immediately following exposure, or after a period of asymptomatic colonization. The immune state at the time of exposure to an infectious agent, interaction between pathogens, and virulence factors intrinsic to the agent are important predictors of an individuals' outcome. Host factors such as extremes of age and underlying disease (e.g. diabetes [56, 57]), human immunodeficiency virus/acquired immune deficiency syndrome [HIV/AIDS] [58, 59], malignancy, and transplants [18, 60, 61] can increase susceptibility to infection as do a variety of medications that alter the normal flora (e.g., antimicrobial agents, gastric acid suppressants, corticosteroids, antirejection drugs, antineoplastic agents, and immunosuppressive drugs). Surgical procedures and radiation therapy impair defenses of the skin and other involved organ systems. Indwelling devices such as urinary catheters, endotracheal tubes, central venous and arterial catheters [62-64] and synthetic implants facilitate development of HAIs by allowing potential pathogens to bypass local defenses that would ordinarily impede their invasion and by providing surfaces for development of bioflms that may facilitate adherence of microorganisms and protect from antimicrobial activity [65]. Some infections associated with invasive procedures result from transmission within the healthcare facility; others arise from the patient's endogenous flora [46-50]. High-risk patient populations with noteworthy risk factors for infection are discussed further in Sections I.D, I.E., and I.F.

I.B.3. Modes of transmission Several classes of pathogens can cause infection, including bacteria, viruses, fungi, parasites, and prions. The modes of transmission vary by type of organism and some infectious agents may be transmitted by more than one route: some are transmitted primarily by direct or indirect contact, (e.g., *Herpes simplex* virus [HSV], respiratory syncytial virus, *Staphylococcus aureus*), others by the droplet, (e.g., influenza virus, *B. pertussis*) or airborne routes (e.g., *M. tuberculosis)*. Other infectious agents, such as bloodborne viruses (e.g., hepatitis B and C viruses [HBV, HCV] and HIV are transmitted rarely in healthcare settings, via percutaneous or mucous membrane exposure. Importantly, not all infectious agents are transmitted from person to person. These are distinguished in Appendix A. The three principal routes of transmission are summarized below.

I.B.3.a*. Contact transmission* The most common mode of transmission, contact transmission is divided into two subgroups: direct contact and indirect contact.

I.B.3.a.i. ***Direct contact transmission*** Direct transmission occurs when microorganisms are transferred from one infected person to another person without a contaminated intermediate object or person. Opportunities for direct contact transmission between patients and healthcare personnel have been summarized in the Guideline for Infection Control in Healthcare Personnel, 1998 [17] and include:

- blood or other blood-containing body fluids from a patient directly enters a caregiver's body through contact with a mucous membrane [66] or breaks (i.e., cuts, abrasions) in the skin [67].
- mites from a scabies-infested patient are transferred to the skin of a caregiver while he/she is having direct ungloved contact with the patient's skin [68, 69].
- a healthcare provider develops herpetic whitlow on a finger after contact with HSV when providing oral care to a patient without using gloves or HSV is transmitted to a patient from a herpetic whitlow on an ungloved hand of a healthcare worker (HCW) [70, 71].

I.B.3.a.ii. ***Indirect contact transmission*** Indirect transmission involves the transfer of an infectious agent through a contaminated intermediate object or person. In the absence of a point-source outbreak, it is difficult to determine how indirect transmission occurs. However, extensive evidence cited in the Guideline for Hand Hygiene in Health-Care Settings suggests that the contaminated hands of healthcare personnel are important contributors to indirect contact transmission [16]. Examples of opportunities for indirect contact transmission include:

- Hands of healthcare personnel may transmit pathogens after touching an infected or colonized body site on one patient or a contaminated inanimate object, if hand hygiene is not performed before touching another patient.[72, 73].
- Patient-care devices (e.g., electronic thermometers, glucose monitoring devices) may transmit pathogens if devices contaminated with blood or body fluids are shared between patients without cleaning and disinfecting between patients[74 75-77].
- Shared toys may become a vehicle for transmitting respiratory viruses (e.g., respiratory syncytial virus [24, 78, 79] or pathogenic bacteria (e.g., *Pseudomonas aeruginosa* [80]) among pediatric patients.
- Instruments that are inadequately cleaned between patients before disinfection or sterilization (e.g., endoscopes or surgical instruments) [81-85] or that have manufacturing defects that interfere with the effectiveness of reprocessing [86, 87] may transmit bacterial and viral pathogens.

Clothing, uniforms, laboratory coats, or isolation gowns used as personal protective equipment (PPE), may become contaminated with potential pathogens after care of a patient colonized or infected with an infectious agent, (e.g., MRSA [88], VRE [89], and *C. difficile* [90]. Although contaminated clothing has not been

implicated directly in transmission, the potential exists for soiled garments to transfer infectious agents to successive patients.

I.B.3.b. ***Droplet transmission*** Droplet transmission is, technically, a form of contact transmission, and some infectious agents transmitted by the droplet route also may be transmitted by the direct and indirect contact routes. However, in contrast to contact transmission, respiratory droplets carrying infectious pathogens transmit infection when they travel directly from the respiratory tract of the infectious individual to susceptible mucosal surfaces of the recipient, generally over short distances, necessitating facial protection. Respiratory droplets are generated when an infected person coughs, sneezes, or talks [91, 92] or during procedures such as suctioning, endotracheal intubation, [93-96], cough induction by chest physiotherapy [97] and cardiopulmonary resuscitation [98, 99]. Evidence for droplet transmission comes from epidemiological studies of disease outbreaks [100-103], experimental studies [104] and from information on aerosol dynamics [91, 105]. Studies have shown that the nasal mucosa, conjunctivae and less frequently the mouth, are susceptible portals of entry for respiratory viruses [106]. The maximum distance for droplet transmission is currently unresolved, although pathogens transmitted by the droplet route have not been transmitted through the air over long distances, in contrast to the airborne pathogens discussed below. Historically, the area of defined risk has been a distance of ≤3 feet around the patient and is based on epidemiologic and simulated studies of selected infections [103, 104]. Using this distance for donning masks has been effective in preventing transmission of infectious agents via the droplet route. However, experimental studies with smallpox [107, 108] and investigations during the global SARS outbreaks of 2003 [101] suggest that droplets from patients with these two infections could reach persons located 6 feet or more from their source. It is likely that the distance droplets travel depends on the velocity and mechanism by which respiratory droplets are propelled from the source, the density of respiratory secretions, environmental factors such as temperature and humidity, and the ability of the pathogen to maintain infectivity over that distance [105]. Thus, a distance of ≤3 feet around the patient is best viewed as an *example* of what is meant by "a short distance from a patient" and should not be used as the sole *criterion* for deciding when a mask should be donned to protect from droplet exposure. Based on these considerations, it may be prudent to don a mask when within 6 to 10 feet of the patient or upon entry into the patient's room, especially when exposure to emerging or highly virulent pathogens is likely. More studies are needed to improve understanding of droplet transmission under various circumstances.

Droplet size is another variable under discussion. Droplets traditionally have been defined as being >5 μm in size. Droplet nuclei, particles arising from desiccation of suspended droplets, have been associated with airborne transmission and defined as ≤5 μm in size [105] , a reflection of the pathogenesis of pulmonary tuberculosis which is not generalizeable to other organisms. Observations of particle dynamics have demonstrated that a range of droplet sizes, including those with diameters of 30μm or greater, can remain suspended

in the air [109] . The behavior of droplets and droplet nuclei affect recommendations for preventing transmission. Whereas fine airborne particles containing pathogens that are able to remain infective may transmit infections over long distances, requiring AIIR to prevent its dissemination within a facility; organisms transmitted by the droplet route do not remain infective over long distances, and therefore do not require special air handling and ventilation. Examples of infectious agents that are transmitted via the droplet route include *Bordetella pertussis* [110], influenza virus [23], adenovirus [111] , rhinovirus [104], *Mycoplasma pneumoniae* [112], SARS-associated coronavirus (SARS-CoV) [21, 96, 113], group A streptococcus [114], and *Neisseria meningitidis* [95, 103, 115]. Although respiratory syncytial virus may be transmitted by the droplet route, direct contact with infected respiratory secretions is the most important determinant of transmission and consistent adherence to Standard plus Contact Precautions prevents transmission in healthcare settings [24, 116, 117].

Rarely, pathogens that are not transmitted routinely by the droplet route are dispersed into the air over short distances. For example, although *S. aureus* is transmitted most frequently by the contact route, viral upper respiratory tract infection has been associated with increased dispersal of *S. aureus* from the nose into the air for a distance of 4 feet under both outbreak and experimental conditions and is known as the “cloud baby” and “cloud adult” phenomenon[118-120].

I.B.3.c. *Airborne transmission* Airborne transmission occurs by dissemination of either airborne droplet nuclei or small particles in the respirable size range containing infectious agents that remain infective over time and distance (e.g., spores of *Aspergillus* spp, and *Mycobacterium tuberculosis*). Microorganisms carried in this manner may be dispersed over long distances by air currents and may be inhaled by susceptible individuals who have not had face-to-face contact with (or been in the same room with) the infectious individual [121-124]. Preventing the spread of pathogens that are transmitted by the airborne route requires the use of special air handling and ventilation systems (e.g., AIIRs) to contain and then safely remove the infectious agent [11, 12]. Infectious agents to which this applies include *Mycobacterium tuberculosis* [124-127], rubeola virus (measles) [122], and varicella-zoster virus (chickenpox) [123]. In addition, published data suggest the possibility that variola virus (smallpox) may be transmitted over long distances through the air under unusual circumstances and AIIRs are recommended for this agent as well; however, droplet and contact routes are the more frequent routes of transmission for smallpox [108, 128, 129]. In addition to AIIRs, respiratory protection with NIOSH certified N95 or higher level respirator is recommended for healthcare personnel entering the AIIR to prevent acquisition of airborne infectious agents such as *M. tuberculosis* [12].

For certain other respiratory infectious agents, such as influenza [130, 131] and rhinovirus [104], and even some gastrointestinal viruses (e.g., norovirus [132] and rotavirus [133]) there is some evidence that the pathogen may be transmitted via small-particle aerosols, under natural and experimental conditions. Such transmission has occurred over distances longer than 3 feet but within a defined

airspace (e.g., patient room), suggesting that it is unlikely that these agents remain viable on air currents that travel long distances. AIIRs are not required routinely to prevent transmission of these agents. Additional issues concerning examples of small particle aerosol transmission of agents that are most frequently transmitted by the droplet route are discussed below.

I.B.3.d. Emerging issues concerning airborne transmission of infectious agents.

I.B.3.d.i. ***Transmission from patients*** The emergence of SARS in 2002, the importation of monkeypox into the United States in 2003, and the emergence of avian influenza present challenges to the assignment of isolation categories because of conflicting information and uncertainty about possible routes of transmission. Although SARS-CoV is transmitted primarily by contact and/or droplet routes, airborne transmission over a limited distance (e.g. within a room), has been suggested, though not proven [134-141]. This is true of other infectious agents such as influenza virus [130] and noroviruses [132, 142, 143]. Influenza viruses are transmitted primarily by close contact with respiratory droplets [23, 102] and acquisition by healthcare personnel has been prevented by Droplet Precautions, even when positive pressure rooms were used in one center [144] However, inhalational transmission could not be excluded in an outbreak of influenza in the passengers and crew of a single aircraft [130]. Observations of a protective effect of UV lights in preventing influenza among patients with tuberculosis during the influenza pandemic of 1957-'58 have been used to suggest airborne transmission [145, 146].
In contrast to the strict interpretation of an airborne route for transmission (i.e., long distances beyond the patient room environment), short distance transmission by small particle aerosols generated under specific circumstances (e.g., during endotracheal intubation) to persons in the immediate area near the patient has been demonstrated. Also, aerosolized particles <100 μm can remain suspended in air when room air current velocities exceed the terminal settling velocities of the particles [109]. SARS-CoV transmission has been associated with endotracheal intubation, noninvasive positive pressure ventilation, and cardio-pulmonary resuscitation [93, 94, 96, 98, 141]. Although the most frequent routes of transmission of noroviruses are contact and food and waterborne routes, several reports suggest that noroviruses may be transmitted through aerosolization of infectious particles from vomitus or fecal material [142, 143, 147, 148]. It is hypothesized that the aerosolized particles are inhaled and subsequently swallowed.

Roy and Milton proposed a new classification for aerosol transmission when evaluating routes of SARS transmission: 1) *obligate*: under natural conditions, disease occurs following transmission of the agent only through inhalation of small particle aerosols (e.g., tuberculosis); 2) *preferential*: natural infection results from transmission through multiple routes, but small particle aerosols are the predominant route (e.g. measles, varicella); and 3) *opportunistic*: agents that naturally cause disease through other routes, but under special circumstances

may be transmitted via fine particle aerosols [149]. This conceptual framework can explain rare occurrences of airborne transmission of agents that are transmitted most frequently by other routes (e.g., smallpox, SARS, influenza, noroviruses). Concerns about unknown or possible routes of transmission of agents associated with severe disease and no known treatment often result in more extreme prevention strategies than may be necessary; therefore, recommended precautions could change as the epidemiology of an emerging infection is defined and controversial issues are resolved.

I.B.3.d.ii. *Transmission from the environment* Some airborne infectious agents are derived from the environment and do not usually involve person-to-person transmission. For example, anthrax spores present in a finely milled powdered preparation can be aerosolized from contaminated environmental surfaces and inhaled into the respiratory tract [150, 151]. Spores of environmental fungi (e.g., *Aspergillus spp*.) are ubiquitous in the environment and may cause disease in immunocompromised patients who inhale aerosolized (e.g., via construction dust) spores [152, 153]. As a rule, neither of these organisms is subsequently transmitted from infected patients. However, there is one well-documented report of person-to-person transmission of *Aspergillus* sp. in the ICU setting that was most likey due to the aerosolization of spores during wound debridement [154]. A Protective Environment refers to isolation practices designed to decrease the risk of exposure to environmental fungal agents in allogeneic HSCT patients [11, 14, 15, 155-158].
Environmental sources of respiratory pathogens (eg. Legionella) transmitted to humans through a common aerosol source is distinct from direct patient-to-patient transmission.

I.B.3.e. Other sources of infection Transmission of infection from sources other than infectious individuals include those associated with *common environmental sources or vehicles* (e.g. contaminated food, water, or medications (e.g. intravenous fluids). Although *Aspergillus* spp. have been recovered from hospital water systems [159], the role of water as a reservoir for immunosuppressed patients remains uncertain. *Vectorborne transmission* of infectious agents from mosquitoes, flies, rats, and other vermin also can occur in healthcare settings. Prevention of vector borne transmission is not addressed in this document.

I.C. Infectious agents of special infection control interest for healthcare settings

Several infectious agents with important infection control implications that either were not discussed extensively in previous isolation guidelines or have emerged recently are discussed below. These are epidemiologically important organisms (e.g., *C. difficile*), agents of bioterrorism, prions, SARS-CoV, monkeypox, noroviruses, and the hemorrhagic fever viruses. Experience with these agents has broadened the understanding of modes of transmission and effective

preventive measures. These agents are included for purposes of information and, for some (i.e., SARS-CoV, monkeypox), because of the lessons that have been learned about preparedness planning and responding effectively to new infectious agents**.**

I.C.1. Epidemiologically important organisms Any infectious agents transmitted in healthcare settings may, under defined conditions, become targeted for control because they are epidemiologically important. *C. difficile* is specifically discussed below because of wide recognition of its current importance in U.S. healthcare facilities. In determining what constitutes an "epidemiologically important organism", the following characteristics apply:

- A propensity for transmission within healthcare facilities based on published reports and the occurrence of temporal or geographic clusters of > 2 patients, (e.g., *C..difficile*, norovirus, respiratory syncytial virus (RSV), influenza, rotavirus, *Enterobacter* spp; *Serratia* spp., group A streptococcus). A single case of healthcare-associated invasive disease caused by certain pathogens (e.g., group A streptococcus post-operatively [160], in burn units [161], or in a LTCF [162]; *Legionella* sp. [14, 163], *Aspergillus* sp. [164]) is generally considered a trigger for investigation and enhanced control measures because of the risk of additional cases and severity of illness associated with these infections. Antimicrobial resistance
- Resistance to first-line therapies (e.g., MRSA, VISA, VRSA, VRE, ESBL-producing organisms).
- Common and uncommon microorganisms with unusual patterns of resistance within a facility (e.g., the first isolate of *Burkholderia cepacia* complex or *Ralstonia* spp. in non-CF patients or a quinolone-resistant strain of *Pseudomonas aeruginosa* in a facility).
- Difficult to treat because of innate or acquired resistance to multiple classes of antimicrobial agents (e.g., *Stenotrophomonas maltophilia, Acinetobacter spp.*).
- Association with serious clinical disease, increased morbidity and mortality (e.g., MRSA and MSSA, group A streptococcus)
- A newly discovered or reemerging pathogen

I.C.1.a. *C.difficile* *C. difficile* is a spore-forming gram positive anaerobic bacillus that was first isolated from stools of neonates in 1935 [165] and identified as the most commonly identified causative agent of antibiotic-associated diarrhea and pseudomembranous colitis in 1977 [166]. This pathogen is a major cause of healthcare-associated diarrhea and has been responsible for many large outbreaks in healthcare settings that were extremely difficult to control. Important factors that contribute to healthcare-associated outbreaks include environmental contamination, persistence of spores for prolonged periods of time, resistance of spores to routinely used disinfectants and antiseptics, hand carriage by healthcare personnel to other patients, and exposure of patients to frequent courses of antimicrobial agents [167] . Antimicrobials most frequently associated

with increased risk of *C. difficile* include third generation cephalosporins, clindamycin, vancomycin, and fluoroquinolones.

Since 2001, outbreaks and sporadic cases of *C. difficile* with increased morbidity and mortality have been observed in several U.S. states, Canada, England and the Netherlands [168-172]. The same strain of *C. difficile* has been implicated in these outbreaks [173]. This strain, toxinotype III, North American PFGE type 1, and PCR-ribotype 027 (NAP1/027). has been found to hyperproduce toxin A (16 fold increase) and toxin B (23 fold increase) compared with isolates from 12 different pulsed-field gel electrophoresisPFGE types. A recent survey of U.S. infectious disease physicians found that 40% perceived recent increases in the incidence and severity of *C. difficile* disease[174]. Standardization of testing methodology and surveillance definitions is needed for accurate comparisons of trends in rates among hospitals [175]. It is hypothesized that the incidence of disease and apparent heightened transmissibility of this new strain may be due, at least in part, to the greater production of toxins A and B, increasing the severity of diarrhea and resulting in more environmental contamination. Considering the greater morbidity, mortality, length of stay, and costs associated with *C. difficile* disease in both acute care and long term care facilities, control of this pathogen is now even more important than previously. Prevention of transmission focuses on syndromic application of Contact Precautions for patients with diarrhea, accurate identification of patients, environmental measures (e.g., rigorous cleaning of patient rooms) and consistent hand hygiene. Use of soap and water, rather than alcohol based handrubs, for mechanical removal of spores from hands, and a bleach-containing disinfectant (5000 ppm) for environmental disinfection, may be valuable when there is transmission in a healthcare facility. See Appendix A for specific recommendations.

I.C.1. b. Multidrug-Resistant Organisms (MDROs) In general, MDROs are defined as microorganisms – predominantly bacteria – that are resistant to one or more classes of antimicrobial agents[176]. Although the names of certain MDROs suggest resistance to only one agent (e.g., methicillin-resistant *Staphylococcus aureus* [MRSA], vancomycin resistant enterococcus [VRE]), these pathogens are usually resistant to all but a few commercially available antimicrobial agents. This latter feature defines MDROs that are considered to be epidemiologically important and deserve special attention in healthcare facilities[177]. Other MDROs of current concern include multidrug-resistant *Streptococcus pneumoniae* (MDRSP) which is resistant to penicillin and other broad-spectrum agents such as macrolides and fluroquinolones, multidrug-resistant gram-negative bacilli (MDR- GNB), especially those producing extended spectrum beta-lactamases (ESBLs); and strains of *S. aureus* that are intermediate or resistant to vancomycin (i.e., VISA and VRSA)[178-197 198].

MDROs are transmitted by the same routes as antimicrobial susceptible infectious agents. Patient-to-patient transmission in healthcare settings, usually via hands of HCWs, has been a major factor accounting for the increase in MDRO incidence and prevalence, especially for MRSA and VRE in acute care

facilities[199-201]. Preventing the emergence and transmission of these pathogens requires a comprehensive approach that includes administrative involvement and measures (e.g., nurse staffing, communication systems, performance improvement processes to ensure adherence to recommended infection control measures), education and training of medical and other healthcare personnel, judicious antibiotic use, comprehensive surveillance for targeted MDROs, application of infection control precautions during patient care, environmental measures (e.g., cleaning and disinfection of the patient care environment and equipment, dedicated single-patient-use of non-critical equipment), and decolonization therapy when appropriate.

The prevention and control of MDROs is a national priority - one that requires that all healthcare facilities and agencies assume responsibility and participate in community-wide control programs[176, 177]. A detailed discussion of this topic and recommendations for prevention was published in 2006 may be found at http://www.cdc.gov/ncidod/dhqp/pdf/ar/mdroGuideline2006.pdf

I.C.2. Agents of bioterrorism CDC has designated the agents that cause anthrax, smallpox, plague, tularemia, viral hemorrhagic fevers, and botulism as Category A (high priority) because these agents can be easily disseminated environmentally and/or transmitted from person to person; can cause high mortality and have the potential for major public health impact; might cause public panic and social disruption; and require special action for public health preparedness[202]. General information relevant to infection control in healthcare settings for Category A agents of bioterrorism is summarized in Table 3. Consult www.bt.cdc.gov for additional, updated Category A agent information as well as information concerning Category B and C agents of bioterrorism and updates. Category B and C agents are important but are not as readily disseminated and cause less morbidity and mortality than Category A agents.

Healthcare facilities confront a different set of issues when dealing with a suspected bioterrorism event as compared with other communicable diseases. An understanding of the epidemiology, modes of transmission, and clinical course of each disease, as well as carefully drafted plans that provide an approach and relevant websites and other resources for disease-specific guidance to healthcare, administrative, and support personnel, are essential for responding to and managing a bioterrorism event. Infection control issues to be addressed include: 1) identifying persons who may be exposed or infected; 2) preventing transmission among patients, healthcare personnel, and visitors; 3) providing treatment, chemoprophylaxis or vaccine to potentially large numbers of people; 4) protecting the environment including the logistical aspects of securing sufficient numbers of AIIRs or designating areas for patient cohorts when there are an insufficient number of AIIRs available;5) providing adequate quantities of appropriate personal protective equipment; and 6) identifying appropriate staff to care for potentially infectious patients (e.g., vaccinated healthcare personnel for

care of patients with smallpox). The response is likely to differ for exposures resulting from an intentional release compared with naturally occurring disease because of the large number persons that can be exposed at the same time and possible differences in pathogenicity.

A variety of sources offer guidance for the management of persons exposed to the most likely agents of bioterrorism. Federal agency websites (e.g., www.usamriid.army.mil/publications/index.html , www.bt.cdc.gov) and state and county health department web sites should be consulted for the most up-to-date information. Sources of information on specific agents include: anthrax [203]; smallpox [204-206]; plague [207, 208]; botulinum toxin [209]; tularemia [210]; and hemorrhagic fever viruses: [211, 212].

I.C.2.a. Pre-event administration of smallpox (vaccinia) vaccine to healthcare personnel Vaccination of personnel in preparation for a possible smallpox exposure has important infection control implications [213-215]. These include the need for meticulous screening for vaccine contraindications in persons who are at increased risk for adverse vaccinia events; containment and monitoring of the vaccination site to prevent transmission in the healthcare setting and at home; and the management of patients with vaccinia-related adverse events [216, 217]. The pre-event U.S. smallpox vaccination program of 2003 is an example of the effectiveness of carefully developed recommendations for both screening potential vaccinees for contraindications and vaccination site care and monitoring. Approximately 760,000 individuals were vaccinated in the Department of Defense and 40,000 in the civilian or public health populations from December 2002 to February 2005, including approximately 70,000 who worked in healthcare settings. There were no cases of eczema vaccinatum, progressive vaccinia, fetal vaccinia, or contact transfer of vaccinia in healthcare settings or in military workplaces [218, 219]. Outside the healthcare setting, there were 53 cases of contact transfer from military vaccinees to close personal contacts (e.g., bed partners or contacts during participation in sports such as wrestling [220]). All contact transfers were from individuals who were not following recommendations to cover their vaccination sites. Vaccinia virus was confirmed by culture or PCR in 30 cases, and two of the confirmed cases resulted from tertiary transfer. All recipients, including one breast-fed infant, recovered without complication. Subsequent studies using viral culture and PCR techniques have confirmed the effectiveness of semipermeable dressings to contain vaccinia [221-224]. This experience emphasizes the importance of ensuring that newly vaccinated healthcare personnel adhere to recommended vaccination-site care, especially if they are to care for high-risk patients. Recommendations for pre-event smallpox vaccination of healthcare personnel and vaccinia-related infection control recommendations are published in the MMWR [216, 225] with updates posted on the CDC bioterrorism web site [205].

I.C.3. Prions Creutzfeldt-Jakob disease (CJD) is a rapidly progressive, degenerative, neurologic disorder of humans with an incidence in the United States of approximately 1 person/million population/year [226, 227]

(www.cdc.gov/ncidod/diseases/cjd/cjd.htm). CJD is believed to be caused by a transmissible proteinaceous infectious agent termed a prion. Infectious prions are isoforms of a host-encoded glycoprotein known as the prion protein. The incubation period (i.e., time between exposure and and onset of symptoms) varies from two years to many decades. However, death typically occurs within 1 year of the onset of symptoms. Approximately 85% of CJD cases occur sporadically with no known environmental source of infection and 10% are familial. Iatrogenic transmission has occurred with most resulting from treatment with human cadaveric pituitary-derived growth hormone or gonadotropin [228, 229], from implantation of contaminated human dura mater grafts [230] or from corneal transplants [231]). Transmission has been linked to the use of contaminated neurosurgical instruments or stereotactic electroencephalogram electrodes [232, 233 , 234 , 235].

Prion diseases in animals include scrapie in sheep and goats, bovine spongiform encephalopathy (BSE, or "mad cow disease") in cattle, and chronic wasting disease in deer and elk [236]. BSE, first recognized in the United Kingdom (UK) in 1986, was associated with a major epidemic among cattle that had consumed contaminated meat and bone meal.

The possible transmission of BSE to humans causing variant CJD (vCJD) was first described in 1996 and subsequently found to be associated with consumption of BSE-contaminated cattle products primarily in the United Kingdom. There is strong epidemiologic and laboratory evidence for a causal association between the causative agent of BSE and vCJD [237]. Although most cases of vCJD have been reported from the UK, a few cases also have been reported from Europe, Japan, Canada, and the United States. Most vCJD cases worldwide lived in or visited the UK during the years of a large outbreak of BSE (1980-96) and may have consumed contaminated cattle products during that time (www.cdc.gov/ncidod/diseases/cjd/cjd.htm). Although there has been no indigenously acquired vCJD in the United States, the sporadic occurrence of BSE in cattle in North America has heightened awareness of the possibility that such infections could occur and have led to increased surveillance activities. Updated information may be found on the following website: www.cdc.gov/ncidod/diseases/cjd/cjd.htm. The public health impact of prion diseases has been reviewed [238].

vCJD in humans has different clinical and pathologic characteristics from sporadic or classic CJD [239], including the following: 1) younger median age at death: 28 (range 16-48) vs. 68 years; 2) longer duration of illness: median 14 months vs. 4-6 months; 3) increased frequency of sensory symptoms and early psychiatric symptoms with delayed onset of frank neurologic signs; and 4) detection of prions in tonsillar and other lymphoid tissues from vCJD patients but not from sporadic CJD patients [240]. Similar to sporadic CJD, there have been no reported cases of direct human-to-human transmission of vCJD by casual or environmental contact, droplet, or airborne routes. Ongoing blood safety surveillance in the U.S. has not detected sporadic CJD transmission through

blood transfusion [241-243]. However, bloodborne transmission of vCJD is believed to have occurred in two UK patients [244, 245]. The following FDA websites provide information on steps that are being taken in the US to protect the blood supply from CJD and vCJD: http://www.fda.gov/cber/gdlns/cjdvcjd.htm;

http://www.fda.gov/cber/gdlns/cjdvcjdq&a.htm.

Standard Precautions are used when caring for patients with suspected or confirmed CJD or vCJD. However, special precautions are recommended for tissue handling in the histology laboratory and for conducting an autopsy, embalming, and for contact with a body that has undergone autopsy [246]. Recommendations for reprocessing surgical instruments to prevent transmission of CJD in healthcare settings have been published by the World Health Organization (WHO) and are currently under review at CDC.

Questions concerning notification of patients potentially exposed to CJD or vCJD through contaminated instruments and blood products from patients with CJD or vCJD or at risk of having vCJD may arise. The risk of transmission associated with such exposures is believed to be extremely low but may vary based on the specific circumstance. Therefore consultation on appropriate options is advised. The United Kingdom has developed several documents that clinicians and patients in the US may find useful (http://www.hpa.org.uk/infections/topics_az/cjd/information_documents.htm).

I.C.4. Severe Acute Respiratory Syndrome (SARS) SARS is a newly discovered respiratory disease that emerged in China late in 2002 and spread to several countries [135, 140]; Mainland China, Hong Kong, Hanoi, Singapore, and Toronto were affected significantly. SARS is caused by SARS CoV, a previously unrecognized member of the coronavirus family [247, 248]. The incubation period from exposure to the onset of symptoms is 2 to 7 days but can be as long as 10 days and uncommonly even longer [249]. The illness is initially difficult to distinguish from other common respiratory infections. Signs and symptoms usually include fever >38.0^{o}C and chills and rigors, sometimes accompanied by headache, myalgia, and mild to severe respiratory symptoms. Radiographic finding of atypical pneumonia is an important clinical indicator of possible SARS. Compared with adults, children have been affected less frequently, have milder disease, and are less likely to transmit SARS-CoV [135, 249-251]. The overall case fatality rate is approximately 6.0%; underlying disease and advanced age increase the risk of mortality (www.who.int/csr/sarsarchive/2003_05_07a/en/).

Outbreaks in healthcare settings, with transmission to large numbers of healthcare personnel and patients have been a striking feature of SARS; undiagnosed, infectious patients and visitors were important initiators of these outbreaks [21, 252-254]. The relative contribution of potential modes of transmission is not precisely known. There is ample evidence for droplet and contact transmission [96, 101, 113]; however, opportunistic airborne transmission cannot be excluded [101, 135-139, 149, 255]. For example, exposure to aerosol-generating

procedures (e.g., endotracheal intubation, suctioning) was associated with transmission of infection to large numbers of healthcare personnel outside of the United States [93, 94, 96, 98, 253].Therefore, aerosolization of small infectious particles generated during these and other similar procedures could be a risk factor for transmission to others within a multi-bed room or shared airspace. A review of the infection control literature generated from the SARS outbreaks of 2003 concluded that the greatest risk of transmission is to those who have close contact, are not properly trained in use of protective infection control procedures, do not consistently use PPE; and that N95 or higher respirators may offer additional protection to those exposed to aerosol- generating procedures and high risk activities [256, 257]. Organizational and individual factors that affected adherence to infection control practices for SARS also were identified [257].

Control of SARS requires a coordinated, dynamic response by multiple disciplines in a healthcare setting [9]. Early detection of cases is accomplished by screening persons with symptoms of a respiratory infection for history of travel to areas experiencing community transmission or contact with SARS patients, followed by implementation of Respiratory Hygiene/Cough Etiquette (i.e., placing a mask over the patient's nose and mouth) and physical separation from other patients in common waiting areas.The precise combination of precautions to protect healthcare personnel has not been determined. At the time of this publication, CDC recommends Standard Precautions, with emphasis on the use of hand hygiene, Contact Precautions with emphasis on environmental cleaning due to the detection of SARS CoV RNA by PCR on surfaces in rooms occupied by SARS patients [138, 254, 258], Airborne Precautions, including use of fit-tested NIOSH-approved N95 or higher level respirators, and eye protection [259]. In Hong Kong, the use of Droplet and Contact Precautions, which included use of a mask but not a respirator, was effective in protecting healthcare personnel[113]. However, in Toronto, consistent use of an N95 respirator was slightly more protective than a mask [93]. It is noteworthy that there was no transmission of SARS-CoV to public hospital workers in Vietnam despite inconsistent use of infection control measures, including use of PPE, which suggests other factors (e.g., severity of disease, frequency of high risk procedures or events, environmental features) may influence opportunities for transmission [260].

SARS-CoV also has been transmitted in the laboratory setting through breaches in recommended laboratory practices. Research laboratories where SARS-CoV was under investigation were the source of most cases reported after the first series of outbreaks in the winter and spring of 2003 [261, 262]. Studies of the SARS outbreaks of 2003 and transmissions that occurred in the laboratory re-affirm the effectiveness of recommended infection control precautions and highlight the importance of consistent adherence to these measures.

Lessons from the SARS outbreaks are useful for planning to respond to future public health crises, such as pandemic influenza and bioterrorism events. Surveillance for cases among patients and healthcare personnel, ensuring availability of adequate supplies and staffing, and limiting access to healthcare

facilities were important factors in the response to SARS that have been summarized [9]. Guidance for infection control precautions in various settings is available at www.cdc.gov/ncidod/sars.

I.C.5. Monkeypox Monkeypox is a rare viral disease found mostly in the rain forest countries of Central and West Africa. The disease is caused by an orthopoxvirus that is similar in appearance to smallpox but causes a milder disease. The only recognized outbreak of human monkeypox in the United States was detected in June 2003 after several people became ill following contact with sick pet prairie dogs. Infection in the prairie dogs was subsequently traced to their contact with a shipment of animals from Africa, including giant Gambian rats [263]. This outbreak demonstrates the importance of recognition and prompt reporting of unusual disease presentations by clinicians to enable prompt identification of the etiology; and the potential of epizootic diseases to spread from animal reservoirs to humans through personal and occupational exposure [264].

Limited data on transmission of monkeypox are available. Transmission from infected animals and humans is believed to occur primarily through direct contact with lesions and respiratory secretions; airborne transmission from animals to humans is unlikely but cannot be excluded, and may have occurred in veterinary practices (e.g., during administration of nebulized medications to ill prairie dogs [265]). Among humans, four instances of monkeypox transmission within hospitals have been reported in Africa among children, usually related to sharing the same ward or bed [266, 267]. Additional recent literature documents transmission of Congo Basin monkeypox in a hospital compound for an extended number of generations [268].

There has been no evidence of airborne or any other person-to-person transmission of monkeypox in the United States, and no new cases of monkeypox have been identified since the outbreak in June 2003 [269]. The outbreak strain is a clade of monkeypox distinct from the Congo Basin clade and may have different epidemiologic properties (including human-to-human transmission potential) from monkeypox strains of the Congo Basin [270]; this awaits further study. Smallpox vaccine is 85% protective against Congo Basin monkeypox [271]. Since there is an associated case fatality rate of $\leq$10%, administration of smallpox vaccine within 4 days to individuals who have had direct exposure to patients or animals with monkeypox is a reasonable consideration [272]. For the most current information on monkeypox, see www.cdc.gov/ncidod/monkeypox/clinicians.htm.

I.C.6. Noroviruses Noroviruses, formerly referred to as Norwalk-like viruses, are members of the *Caliciviridae* family. These agents are transmitted via contaminated food or water and from person-to-person, causing explosive outbreaks of gastrointestinal disease [273]. Environmental contamination also has been documented as a contributing factor in ongoing transmission during outbreaks [274, 275]. Although noroviruses cannot be propagated in cell culture,

DNA detection by molecular diagnostic techniques has facilitated a greater appreciation of their role in outbreaks of gastrointestinal disease [276]. Reported outbreaks in hospitals [132, 142, 277], nursing homes [275, 278-283], cruise ships [284, 285], hotels [143, 147], schools [148], and large crowded shelters established for hurricane evacuees [286], demonstrate their highly contagious nature, the disruptive impact they have in healthcare facilities and the community, and the difficulty of controlling outbreaks in settings where people share common facilites and space. Of note, there is nearly a 5 fold increase in the risk to patients in outbreaks where a patient is the index case compared with exposure of patients during outbreaks where a staff member is the index case [287].

The average incubation period for gastroenteritis caused by noroviruses is 12-48 hours and the clinical course lasts 12-60 hours [273]. Illness is characterized by acute onset of nausea, vomiting, abdominal cramps, and/or diarrhea. The disease is largely self-limited; rarely, death caused by severe dehydration can occur, particularly among the elderly with debilitating health conditions.

The epidemiology of norovirus outbreaks shows that even though primary cases may result from exposure to a fecally-contaminated food or water, secondary and tertiary cases often result from person-to-person transmission that is facilitated by contamination of fomites [273, 288] and dissemination of infectious particles, especially during the process of vomiting [132, 142, 143, 147, 148, 273, 279, 280]. Widespread, persistent and inapparent contamination of the environment and fomites can make outbreaks extremely difficult to control [147, 275, 284].These clinical observations and the detection of norovirus DNA on horizontal surfaces 5 feet above the level that might be touched normally suggest that, under certain circumstances, aerosolized particles may travel distances beyond 3 feet [147]. It is hypothesized that infectious particles may be aerosolized from vomitus, inhaled, and swallowed. In addition, individuals who are responsible for cleaning the environment may be at increased risk of infection. Development of disease and transmission may be facilitated by the low infectious dose (i.e., <100 viral particles) [289] and the resistance of these viruses to the usual cleaning and disinfection agents (i.e., may survive $\leq$ 10 ppm chlorine) [290-292]. An alternate phenolic agent that was shown to be effective against feline calicivirus was used for environmental cleaning in one outbreak [275, 293]. There are insufficient data to determine the efficacy of alcohol-based hand rubs against noroviruses when the hands are not visibly soiled [294]. Absence of disease in certain individuals during an outbreak may be explained by protection from infection conferred by the B histo-blood group antigen [295]. Consultation on outbreaks of gastroenteritis is available through CDC's Division of Viral and Rickettsial Diseases [296].

I.C.7. Hemorrhagic fever viruses (HFV) The hemorrhagic fever viruses are a mixed group of viruses that cause serious disease with high fever, skin rash, bleeding diathesis, and in some cases, high mortality; the disease caused is referred to as viral hemorrhagic fever (VHF). Among the more commonly known HFVs are Ebola and Marburg viruses (Filoviridae), Lassa virus (Arenaviridae), Crimean-Congo hemorrhagic fever and Rift Valley Fever virus (Bunyaviridae),

and Dengue and Yellow fever viruses (Flaviviridae) [212, 297]. These viruses are transmitted to humans via contact with infected animals or via arthropod vectors. While none of these viruses is endemic in the United States, outbreaks in affected countries provide potential opportunities for importation by infected humans and animals. Furthermore, there are concerns that some of these agents could be used as bioweapons [212]. Person-to-person transmission is documented for Ebola, Marburg, Lassa and Crimean-Congo hemorrhagic fever viruses. In resource-limited healthcare settings, transmission of these agents to healthcare personnel, patients and visitors has been described and in some outbreaks has accounted for a large proportion of cases [298-300]. Transmissions within households also have occurred among individuals who had direct contact with ill persons or their body fluids, but not to those who did not have such contact [301].

Evidence concerning the transmission of HFVs has been summarized [212, 302]. Person-to-person transmission is associated primarily with direct blood and body fluid contact. Percutaneous exposure to contaminated blood carries a particularly high risk for transmission and increased mortality [303, 304]. The finding of large numbers of Ebola viral particles in the skin and the lumina of sweat glands has raised concern that transmission could occur from direct contact with intact skin though epidemiologic evidence to support this is lacking [305]. Postmortem handling of infected bodies is an important risk for transmission [301, 306, 307]. In rare situations, cases in which the mode of transmission was unexplained among individuals with no known direct contact , have led to speculation that airborne transmission could have occurred [298]. However, airborne transmission of naturally occurring HFVs in humans has not been seen. In one study of airplane passengers exposed to an in-flight index case of Lassa fever, there was no transmission to any passengers[308].

In the laboratory setting, animals have been infected experimentally with Marburg or Ebola viruses via direct inoculation of the nose, mouth and/or conjunctiva [309, 310] and by using mechanically generated virus-containing aerosols [311, 312]. Transmission of Ebola virus among laboratory primates in an animal facility has been described [313]. Secondarily infected animals were in individual cages and separated by approximately 3 meters. Although the possibility of airborne transmission was suggested, the authors were not able to exclude droplet or indirect contact transmission in this incidental observation.

Guidance on infection control precautions for HVFs that are transmitted person-to-person have been published by CDC [1, 211] and by the Johns Hopkins Center for Civilian Biodefense Strategies [212]. The most recent recommendations at the time of publication of this document were posted on the CDC website on 5/19/05 [314]. Inconsistencies among the various recommendations have raised questions about the appropriate precautions to use in U.S. hospitals. In less developed countries, outbreaks of HFVs have been controlled with basic hygiene, barrier precautions, safe injection practices, and safe burial practices [299, 306]. The preponderance of evidence on HFV transmission indicates that Standard, Contact and Droplet Precautions with eye protection are effective in protecting

healthcare personnel and visitors who may attend an infected patient. Single gloves are adequate for routine patient care; double-gloving is advised during invasive procedures (e.g., surgery) that pose an increased risk for blood exposure. Routine eye protection (i.e. goggles or face shield) is particularly important. Fluid-resistant gowns should be worn for all patient contact. Airborne Precautions are not required for routine patient care; however, use of AIIRs is prudent when procedures that could generate infectious aerosols are performed (e.g., endotracheal intubation, bronchoscopy, suctioning, autopsy procedures involving oscillating saws). N95 or higher level respirators may provide added protection for individuals in a room during aerosol-generating procedures (Table 3, Appendix A). When a patient with a syndrome consistent with hemorrhagic fever also has a history of travel to an endemic area, precautions are initiated upon presentation and then modified as more information is obtained (Table 2). Patients with hemorrhagic fever syndrome in the setting of a suspected bioweapon attack should be managed using Airborne Precautions, including AIIRs, since the epidemiology of a potentially weaponized hemorrhagic fever virus is unpredictable.

I.D. Transmission risks associated with specific types of healthcare settings

Numerous factors influence differences in transmission risks among the various healthcare settings. These include the population characteristics (e.g., increased susceptibility to infections, type and prevalence of indwelling devices), intensity of care, exposure to environmental sources, length of stay, and frequency of interaction between patients/residents with each other and with HCWs. These factors, as well as organizational priorities, goals, and resources, influence how different healthcare settings adapt transmission prevention guidelines to meet their specific needs [315, 316]. Infection control management decisions are informed by data regarding institutional experience/epidemiology, trends in community and institutional HAIs, local, regional, and national epidemiology, and emerging infectious disease threats.

I.D.1. Hospitals Infection transmission risks are present in all hospital settings. However, certain hospital settings and patient populations have unique conditions that predispose patients to infection and merit special mention. These are often sentinel sites for the emergence of new transmission risks that may be unique to that setting or present opportunities for transmission to other settings in the hospital.

I.D.1.a. Intensive Care Units Intensive care units (ICUs) serve patients who are immunocompromised by disease state and/or by treatment modalities, as well as patients with major trauma, respiratory failure and other life-threatening conditions (e.g., myocardial infarction, congestive heart failure, overdoses, strokes, gastrointestinal bleeding, renal failure, hepatic failure, multi-organ system failure, and the extremes of age). Although ICUs account for a relatively

small proportion of hospitalized patients, infections acquired in these units accounted for >20% of all HAIs [317]. In the National Nosocomial Infection Surveillance (NNIS) system, 26.6% of HAIs were reported from ICU and high risk nursery (NICU) patients in 2002 (NNIS, unpublished data). This patient population has increased susceptibility to colonization and infection, especially with MDROs and *Candida* sp. [318, 319], because of underlying diseases and conditions, the invasive medical devices and technology used in their care (e.g. central venous catheters and other intravascular devices, mechanical ventilators, extracorporeal membrane oxygenation (ECMO), hemodialysis/-filtration, pacemakers, implantable left ventricular assist devices), the frequency of contact with healthcare personnel, prolonged length of stay, and prolonged exposure to antimicrobial agents [320-331]. Furthermore, adverse patient outcomes in this setting are more severe and are associated with a higher mortality [332]. Outbreaks associated with a variety of bacterial, fungal and viral pathogens due to common-source and person-to-person transmissions are frequent in adult and pediatric ICUs [31, 333-336, 337 , 338] .

I.D.1.b. Burn Units Burn wounds can provide optimal conditions for colonization, infection, and transmission of pathogens; infection acquired by burn patients is a frequent cause of morbidity and mortality [320, 339, 340]. In patients with a burn injury involving ≥30% of the total body surface area (TBSA), the risk of invasive burn wound infection is particularly high [341, 342]. Infections that occur in patients with burn injury involving <30% TBSA are usually associated with the use of invasive devices. Methicillin-susceptible *Staphylococcus aureus*, MRSA, enterococci, including VRE, gram-negative bacteria, and candida are prevalent pathogens in burn infections [53, 340, 343-350] and outbreaks of these organisms have been reported [351-354]. Shifts over time in the predominance of pathogens causing infections among burn patients often lead to changes in burn care practices [343, 355-358]. Burn wound infections caused by *Aspergillus* sp. or other environmental molds may result from exposure to supplies contaminated during construction [359] or to dust generated during construction or other environmental disruption [360].

Hydrotherapy equipment is an important environmental reservoir of gram-negative organisms. Its use for burn care is discouraged based on demonstrated associations between use of contaminated hydrotherapy equipment and infections. Burn wound infections and colonization, as well as bloodstream infections, caused by multidrug-resistant *P. aeruginosa* [361], *A. baumannii* [362], and MRSA [352] have been associated with hydrotherapy; excision of burn wounds in operating rooms is preferred.

Advances in burn care, specifically early excision and grafting of the burn wound, use of topical antimicrobial agents, and institution of early enteral feeding, have led to decreased infectious complications. Other advances have included prophylactic antimicrobial usage, selective digestive decontamination (SDD), and use of antimicrobial-coated catheters (ACC), but few epidemiologic studies and no efficacy studies have been performed to show the relative benefit of these measures [357].

There is no consensus on the most effective infection control practices to prevent transmission of infections to and from patients with serious burns (e.g., single-bed rooms [358], laminar flow [363] and high efficiency particulate air filtration [HEPA] [360] or maintaining burn patients in a separate unit without exposure to patients or equipment from other units [364]). There also is controversy regarding the need for and type of barrier precautions for routine care of burn patients. One retrospective study demonstrated efficacy and cost effectiveness of a simplified barrier isolation protocol for wound colonization, emphasizing handwashing and use of gloves, caps, masks and plastic impermeable aprons (rather than isolation gowns) for direct patient contact [365]. However, there have been no studies that define the most effective combination of infection control precautions for use in burn settings. Prospective studies in this area are needed.

I.D.1.c. Pediatrics Studies of the epidemiology of HAIs in children have identified unique infection control issues in this population [63, 64, 366-370]. Pediatric intensive care unit (PICU) patients and the lowest birthweight babies in the high-risk nursery (HRN) monitored in the NNIS system have had high rates of central venous catheter-associated bloodstream infections [64, 320, 369-372]. Additionally, there is a high prevalence of community-acquired infections among hospitalized infants and young children who have not yet become immune either by vaccination or by natural infection. The result is more patients and their sibling visitors with transmissible infections present in pediatric healthcare settings, especially during seasonal epidemics (e.g., pertussis [36, 40, 41], respiratory viral infections including those caused by RSV [24], influenza viruses [373], parainfluenza virus [374], human metapneumovirus [375], and adenoviruses [376]; rubeola [measles] [34], varicella [chickenpox] [377], and rotavirus [38, 378]).

Close physical contact between healthcare personnel and infants and young children (eg. cuddling, feeding, playing, changing soiled diapers, and cleaning copious uncontrolled respiratory secretions) provides abundant opportunities for transmission of infectious material. Practices and behaviors such as congregation of children in play areas where toys and bodily secretions are easily shared and family members rooming-in with pediatric patients can further increase the risk of transmission. Pathogenic bacteria have been recovered from toys used by hospitalized patients [379]; contaminated bath toys were implicated in an outbreak of multidrug-resistant *P. aeruginosa* on a pediatric oncology unit [80]. In addition, several patient factors increase the likelihood that infection will result from exposure to pathogens in healthcare settings (e.g., immaturity of the neonatal immune system, lack of previous natural infection and resulting immunity, prevalence of patients with congenital or acquired immune deficiencies, congenital anatomic anomalies, and use of life-saving invasive devices in neontal and pediatric intensive care units) [63]. There are theoretical concerns that infection risk will increase in association with innovative practices used in the NICU for the purpose of improving developmental outcomes, Such factors include co-bedding [380] and kangaroo care [381] that may increase opportunity for skin-to-skin exposure of multiple gestation infants to each other and to their mothers, respectively; although infection risk smay actually be

reduced among infants receiving kangaroo care [382]. Children who attend child care centers [383, 384] and pediatric rehabilitation units [385] may increase the overall burden of antimicrobial resistance (eg. by contributing to the reservoir of community-associated MRSA [CA-MRSA]) [386-391]. Patients in chronic care facilities may have increased rates of colonization with resistant GNBs and may be sources of introduction of resistant organisms to acute care settings [50].

I.D.2. Nonacute healthcare settings Healthcare is provided in various settings outside of hospitals including facilities, such as long-term care facilities (LTCF) (e.g. nursing homes), homes for the developmentally disabled, settings where behavioral health services are provided, rehabilitation centers and hospices[392]. In addition, healthcare may be provided in nonhealthcare settings such as workplaces with occupational health clinics, adult day care centers, assisted living facilities, homeless shelters, jails and prisons, school clinics and infirmaries. Each of these settings has unique circumstances and population risks to consider when designing and implementing an infection control program. Several of the most common settings and their particular challenges are discussed below. While this Guideline does not address each setting, the principles and strategies provided may be adapted and applied as appropriate.

I.D.2.a. Long-term care The designation LTCF applies to a diverse group of residential settings, ranging from institutions for the developmentally disabled to nursing homes for the elderly and pediatric chronic-care facilities [393-395]. Nursing homes for the elderly predominate numerically and frequently represent long-term care as a group of facilities. Approximately 1.8 million Americans reside in the nation's 16,500 nursing homes [396]. Estimates of HAI rates of 1.8 to 13.5 per 1000 resident-care days have been reported with a range of 3 to 7 per 1000 resident-care days in the more rigorous studies [397-401]. The infrastructure described in the Department of Veterans Affairs nursing home care units is a promising example for the development of a nationwide HAI surveillance system for LTCFs [402].

LCTFs are different from other healthcare settings in that elderly patients at increased risk for infection are brought together in one setting and remain in the facility for extended periods of time; for most residents, it is their home. An atmosphere of community is fostered and residents share common eating and living areas, and participate in various facility-sponsored activities [403, 404]. Since able residents interact freely with each other, controlling transmission of infection in this setting is challenging [405]. Residents who are colonized or infected with certain microorganisms are, in some cases, restricted to their room. However, because of the psychosocial risks associated with such restriction, it has been recommended that psychosocial needs be balanced with infection control needs in the LTCF setting [406-409]. Documented LTCF outbreaks have been caused by various viruses (e.g., influenza virus [35, 410-412], rhinovirus [413], adenovirus (conjunctivitis) [414], norovirus [278, 279 275, 281]) and bacteria, including group A streptococcus [162], *B. pertussis* [415], non-susceptible *S. pneumoniae* [197, 198], other MDROs, and *Clostridium difficile* [416]) These pathogens can lead to substantial

morbidity and mortality, and increased medical costs; prompt detection and implementation of effective control measures are required.

Risk factors for infection are prevalent among LTCF residents [395, 417, 418]. Age-related declines in immunity may affect responses to immunizations for influenza and other infectious agents, and increase susceptibility to tuberculosis. Immobility, incontinence, dysphagia, underlying chronic diseases, poor functional status, and age-related skin changes increase susceptibility to urinary, respiratory and cutaneous and soft tissue infections, while malnutrition can impair wound healing [419-423]. Medications (e.g., drugs that affect level of consciousness, immune function, gastric acid secretions, and normal flora, including antimicrobial therapy) and invasive devices (e.g., urinary catheters and feeding tubes) heighten susceptibility to infection and colonization in LTCF residents [424-426]. Finally, limited functional status and total dependence on healthcare personnel for activities of daily living have been identified as independent risk factors for infection [401, 417, 427] and for colonization with MRSA [428, 429] and ESBL-producing *K. pneumoniae* [430]. Several position papers and review articles have been published that provide guidance on various aspects of infection control and antimicrobial resistance in LTCFs [406-408, 431-436]. The Centers for Medicare and Medicaid Services (CMS) have established regulations for the prevention of infection in LTCFs [437].

Because residents of LTCFs are hospitalized frequently, they can transfer pathogens between LTCFs and healthcare facilities in which they receive care [8, 438-441]. This is also true for pediatric long-term care populations. Pediatric chronic care facilities have been associated with importing extended-spectrum cephalosporin-resistant, gram-negative bacilli into one PICU [50]. Children from pediatric rehabilitation units may contribute to the reservoir of community-associated MRSA [385, 389-391].

I.D.2.b. Ambulatory Care In the past decade, healthcare delivery in the United States has shifted from the acute, inpatient hospital to a variety of ambulatory and community-based settings, including the home. Ambulatory care is provided in hospital-based outpatient clinics, nonhospital-based clinics and physician offices, public health clinics, free-standing dialysis centers, ambulatory surgical centers, urgent care centers, and many others. In 2000, there were 83 million visits to hospital outpatient clinics and more than 823 million visits to physician offices [442]; ambulatory care now accounts for most patient encounters with the health care system [443]. In these settings, adapting transmission prevention guidelines is challenging because patients remain in common areas for prolonged periods waiting to be seen by a healthcare provider or awaiting admission to the hospital, examination or treatment rooms are turned around quickly with limited cleaning, and infectious patients may not be recognized immediately. Furthermore, immunocompromised patients often receive chemotherapy in infusion rooms where they stay for extended periods of time along with other types of patients.

There are few data on the risk of HAIs in ambulatory care settings, with the exception of hemodialysis centers [18, 444, 445]. Transmission of infections in outpatient settings has been reviewed in three publications [446-448]. Goodman and Solomon summarized 53 clusters of infections associated with the outpatient setting from 1961-1990 [446]. Overall, 29 clusters were associated with common source transmission from contaminated solutions or equipment, 14 with person-to-person transmission from or involving healthcare personnel and ten associated with airborne or droplet transmission among patients and healthcare workers. Transmission of bloodborne pathogens (i.e., hepatitis B and C viruses and, rarely, HIV) in outbreaks, sometimes involving hundreds of patients, continues to occur in ambulatory settings. These outbreaks often are related to common source exposures, usually a contaminated medical device, multi-dose vial, or intravenous solution [82, 449-453]. In all cases, transmission has been attributed to failure to adhere to fundamental infection control principles, including safe injection practices and aseptic technique.This subject has been reviewed and recommended infection control and safe injection practices summarized [454].

Airborne transmission of *M.tuberculosis* and measles in ambulatory settings, most frequently emergency departments, has been reported [34, 127, 446, 448, 455-457]. Measles virus was transmitted in physician offices and other outpatient settings during an era when immunization rates were low and measles outbreaks in the community were occurring regularly [34, 122, 458]. Rubella has been transmitted in the outpatient obstetric setting [33]; there are no published reports of varicella transmission in the outpatient setting. In the ophthalmology setting, adenovirus type 8 epidemic keratoconjunctivitis has been transmitted via incompletely disinfected ophthalmology equipment and/or from healthcare workers to patients, presumably by contaminated hands [17, 446, 448, 459-462].

If transmission in outpatient settings is to be prevented, screening for potentially infectious symptomatic and asymptomatic individuals, especially those who may be at risk for transmitting airborne infectious agents (e.g., *M. tuberculosis,* varicella-zoster virus, rubeola [measles]), is necessary at the start of the initial patient encounter. Upon identification of a potentially infectious patient, implementation of prevention measures, including prompt separation of potentially infectious patients and implementation of appropriate control measures (e.g., Respiratory Hygiene/Cough Etiquette and Transmission-Based Precautions) can decrease transmission risks [9, 12]. Transmission of MRSA and VRE in outpatient settings has not been reported, but the association of CA-MRSA in healthcare personnel working in an outpatient HIV clinic with environmental CA-MRSA contamination in that clinic, suggests the possibility of transmission in that setting [463]. Patient-to-patient transmission of *Burkholderia species* and *Pseudomonas aeruginosa* in outpatient clinics for adults and children with cystic fibrosis has been confirmed [464, 465].

I.D.2.c. Home Care Home care in the United States is delivered by over 20,000 provider agencies that include home health agencies, hospices, durable medical equipment providers, home infusion therapy services, and personal care and

support services providers. Home care is provided to patients of all ages with both acute and chronic conditions. The scope of services ranges from assistance with activities of daily living and physical and occupational therapy to the care of wounds, infusion therapy, and chronic ambulatory peritoneal dialysis (CAPD).

The incidence of infection in home care patients, other than those associated with infusion therapy is not well studied [466-471]. However, data collection and calculation of infection rates have been accomplished for central venous catheter-associated bloodstream infections in patients receiving home infusion therapy [470-474] and for the risk of blood contact through percutaneous or mucosal exposures, demonstrating that surveillance can be performed in this setting [475]. Draft definitions for home care associated infections have been developed [476].

Transmission risks during home care are presumed to be minimal. The main transmission risks to home care patients are from an infectious healthcare provider or contaminated equipment; providers also can be exposed to an infectious patient during home visits. Since home care involves patient care by a limited number of personnel in settings without multiple patients or shared equipment, the potential reservoir of pathogens is reduced. Infections of home care providers, that could pose a risk to home care patients include infections transmitted by the airborne or droplet routes (e.g., chickenpox, tuberculosis, influenza), and skin infestations (e.g., scabies [69] and lice) and infections (e.g.,impetigo) transmitted by direct or indirect contact. There are no published data on indirect transmission of MDROs from one home care patient to another, although this is theoretically possible if contaminated equipment is transported from an infected or colonized patient and used on another patient. Of note, investigation of the first case of VISA in homecare [186] and the first 2 reported cases of VRSA [178, 180, 181, 183] found no evidence of transmission of VISA or VRSA to other home care recipients. Home health care also may contribute to antimicrobial resistance; a review of outpatient vancomycin use found 39% of recipients did not receive the antibiotic according to recommended guidelines [477].

Although most home care agencies implement policies and procedures to prevent transmission of organisms, the current approach is based on the adaptation of the *1996 Guideline for Isolation Precautions in Hospitals* [1] as well as other professional guidance [478, 479]. This issue has been very challenging in the home care industry and practice has been inconsistent and frequently not evidence-based. For example, many home health agencies continue to observe "nursing bag technique," a practice that prescribes the use of barriers between the nursing bag and environmental surfaces in the home [480]. While the home environment may not always appear clean, the use of barriers between two non-critical surfaces has been questioned [481, 482]. Opportunites exist to conduct research in home care related to infection transmission risks [483].

I.D.2.d. Other sites of healthcare delivery Facilities that are not primarily healthcare settings but in which healthcare is delivered include clinics in correctional facilities and shelters. Both settings can have suboptimal features,

such as crowded conditions and poor ventilation. Economically disadvantaged individuals who may have chronic illnesses and healthcare problems related to alcoholism, injection drug use, poor nutrition, and/or inadequate shelter often receive their primary healthcare at sites such as these [484]. Infectious diseases of special concern for transmission include tuberculosis, scabies, respiratory infections (e.g., *N. meningitides, S. pneumoniae*), sexually transmitted and bloodborne diseases (e.g.,HIV, HBV, HCV, syphilis, gonorrhea), hepatitis A virus (HAV), diarrheal agents such as norovirus, and foodborne diseases [286, 485-488]. A high index of suspicion for tuberculosis and CA-MRSA in these populations is needed as outbreaks in these settings or among the populations they serve have been reported [489-497].

Patient encounters in these types of facilities provide an opportunity to deliver recommended immunizations and screen for *M. tuberculosis* infection in addition to diagnosing and treating acute illnesses [498]. Recommended infection control measures in these non-traditional areas designated for healthcare delivery are the same as for other ambulatory care settings. Therefore, these settings must be equipped to observe Standard Precautions and, when indicated, Transmission-based Precautions.

I.E. Transmission risks associated with special patient populations

As new treatments emerge for complex diseases, unique infection control challenges associated with special patient populations need to be addressed.

I.E.1. Immunocompromised patients Patients who have congenital primary immune deficiencies or acquired disease (eg. treatment-induced immune deficiencies) are at increased risk for numerous types of infections while receiving healthcare and may be located throughout the healthcare facility. The specific defects of the immune system determine the types of infections that are most likely to be acquired (e.g., viral infections are associated with T-cell defects and fungal and bacterial infections occur in patients who are neutropenic). As a general group, immunocompromised patients can be cared for in the same environment as other patients; however, it is always advisable to minimize exposure to other patients with transmissible infections such as influenza and other respiratory viruses [499, 500]. The use of more intense chemotherapy regimens for treatment of childhood leukemia may be associated with prolonged periods of neutropenia and suppression of other components of the immune system, extending the period of infection risk and raising the concern that additional precautions may be indicated for select groups [501, 502]. With the application of newer and more intense immunosuppressive therapies for a variety of medical conditions (e.g., rheumatologic disease [503, 504], inflammatory bowel disease [505]), immunosuppressed patients are likely to be more widely distributed throughout a healthcare facility rather than localized to single patient units (e.g.

hematology-oncology). Guidelines for preventing infections in certain groups of immunocompromised patients have been published [15, 506, 507].

Published data provide evidence to support placing allogeneic HSCT patients in a Protective Environment [15, 157, 158]. Also, three guidelines have been developed that address the special requirements of these immunocompromised patients, including use of antimicrobial prophylaxis and engineering controls to create a Protective Environment for the prevention of infections caused by *Aspergillus* spp. and other environmental fungi [11, 14, 15]. As more intense chemotherapy regimens associated with prolonged periods of neutropenia or graft-versus-host disease are implemented, the period of risk and duration of environmental protection may need to be prolonged beyond the traditional 100 days [508].

I.E.2. Cystic fibrosis patients Patients with cystic fibrosis (CF) require special consideration when developing infection control guidelines. Compared to other patients, CF patients require additional protection to prevent transmission from contaminated respiratory therapy equipment [509-513]. Infectious agents such as *Burkholderia cepacia* complex and *P. aeruginosa* [464, 465, 514, 515] have unique clinical and prognostic significance. In CF patients, *B. cepacia* infection has been associated with increased morbidity and mortality [516-518], while delayed acquisition of chronic *P.aeruginosa* infection may be associated with an improved long-term clinical outcome [519, 520].

Person-to-person transmission of *B. cepacia* complex has been demonstrated among children [517] and adults [521] with CF in healthcare settings [464, 522], during various social contacts [523], most notably attendance at camps for patients with CF [524], and among siblings with CF [525]. Successful infection control measures used to prevent transmission of respiratory secretions include segregation of CF patients from each other in ambulatory and hospital settings (including use of private rooms with separate showers), environmental decontamination of surfaces and equipment contaminated with respiratory secretions, elimination of group chest physiotherapy sessions, and disbanding of CF camps [97, 526]. The Cystic Fibrosis Foundation published a consensus document with evidence-based recommendations for infection control practices for CF patients [20].

I.F. New therapies associated with potentially transmissible infectious agents

I.F.1. Gene therapy Gene therapy has has been attempted using a number of different viral vectors, including nonreplicating retroviruses, adenoviruses, adeno-associated viruses, and replication-competent strains of poxviruses. Unexpected adverse events have restricted the prevalence of gene therapy protocols.

The infectious hazards of gene therapy are theoretical at this time, but require meticulous surveillance due to the possible occurrence of in vivo recombination

and the subsequent emergence of a transmissible genetically altered pathogen. Greatest concern attends the use of replication-competent viruses, especially vaccinia. As of the time of publication, no reports have described transmission of a vector virus from a gene therapy recipient to another individual, but surveillance is ongoing. Recommendations for monitoring infection control issues throughout the course of gene therapy trials have been published [527-529].

I.F.2. Infections transmitted through blood, organs and other tissues The potential hazard of transmitting infectious pathogens through biologic products is a small but ever present risk, despite donor screening. Reported infections transmitted by transfusion or transplantation include West Nile Virus infection [530] cytomegalovirus infection [531], Creutzfeldt-Jacob disease [230], hepatitis C [532], infections with *Clostridium* spp. [533] and group A streptococcus [534], malaria [535], babesiosis [536], Chagas disease [537], lymphocytic choriomeningitis [538], and rabies [539, 540]. Therefore, it is important to consider receipt of biologic products when evaluating patients for potential sources of infection.

I.F.3. Xenotransplantation The transplantation of nonhuman cells, tissues, and organs into humans potentially exposes patients to zoonotic pathogens. Transmission of known zoonotic infections (e.g., trichinosis from porcine tissue), constitutes one concern, but also of concern is the possibility that transplantation of nonhuman cells, tissues, or organs may transmit previously unknown zoonotic infections (xenozoonoses) to immunosuppressed human recipients. Potential infections that might accompany transplantation of porcine organs have been described [541]. Guidelines from the U.S. Public Health Service address many infectious diseases and infection control issues that surround the developing field of xenotransplantation [542]); work in this area is ongoing.

Part II:

Fundamental elements needed to prevent transmission of infectious agents in healthcare settings

II.A. Healthcare system components that influence the effectiveness of precautions to prevent transmission

II.A.1. Administrative measures Healthcare organizations can demonstrate a commitment to preventing transmission of infectious agents by incorporating infection control into the objectives of the organization's patient and occupational safety programs [543-547]. An infrastructure to guide, support, and monitor adherence to Standard and Transmission-Based Precautions [434, 548, 549] will facilitate fulfillment of the organization's mission and achievement of the Joint Commission on Accreditation of Healthcare Organization's patient safety goal to decrease HAIs [550]. Policies and procedures that explain how Standard and Transmission-Based Precautions are applied, including systems used to identify and communicate information about patients with potentially transmissible infectious agents, are essential to ensure the success of these measures and may vary according to the characteristics of the organization.

A key administrative measure is provision of fiscal and human resources for maintaining infection control and occupational health programs that are responsive to emerging needs. Specific components include bedside nurse [551] and infection prevention and control professional (ICP) staffing levels [552], inclusion of ICPs in facility construction and design decisions [11], clinical microbiology laboratory support [553, 554], adequate supplies and equipment including facility ventilation systems [11], adherence monitoring [555], assessment and correction of system failures that contribute to transmission [556, 557], and provision of feedback to healthcare personnel and senior administrators [434, 548, 549, 558]. The positive influence of institutional leadership has been demonstrated repeatedly in studies of HCW adherence to recommended hand hygiene practices [176, 177, 434, 548, 549, 559-564]. Healthcare administrator involvement in infection control processes can improve administrators' awareness of the rationale and resource requirements for following recommended infection control practices.

Several administrative factors may affect the transmission of infectious agents in healthcare settings: institutional culture, individual worker behavior, and the work environment. Each of these areas is suitable for performance improvement monitoring and incorporation into the organization's patient safety goals [543, 544, 546, 565].

II.A.1.a.Scope of work and staffing needs for infection control professionals

The effectiveness of infection surveillance and control programs in preventing nosocomial infections in United States hospitals was assessed by the CDC through the Study on the Efficacy of Nosocomial Infection Control (SENIC Project) conducted 1970-76 [566]. In a representative sample of US general hospitals, those with a trained infection control physician or microbiologist involved in an infection control program, and at least one infection control nurse per 250 beds, were associated with a 32% lower rate of four infections studied (CVC-associated bloodstream infections, ventilator-associated pneumonias, catheter-related urinary tract infections, and surgical site infections).

Since that landmark study was published, responsibilities of ICPs have expanded commensurate with the growing complexity of the healthcare system, the patient populations served, and the increasing numbers of medical procedures and devices used in all types of healthcare settings. The scope of work of ICPs was first assessed in 1982 [567-569] by the Certification Board of Infection Control (CBIC), and has been re-assessed every five years since that time [558, 570-572]. The findings of these task analyses have been used to develop and update the Infection Control Certification Examination, offered for the first time in 1983. With each survey, it is apparent that the role of the ICP is growing in complexity and scope, beyond traditional infection control activities in acute care hospitals. Activities currently assigned to ICPs in response to emerging challenges include: 1) surveillance and infection prevention at facilities other than acute care hospitals e.g., ambulatory clinics, day surgery centers, long term care facilities, rehabilitation centers, home care; 2) oversight of employee health services related to infection prevention, e.g. assessment of risk and administration of recommended treatment following exposure to infectious agents, tuberculosis screening, influenza vaccination, respiratory protection fit testing, and administration of other vaccines as indicated, such as smallpox vaccine in 2003; 3) preparedness planning for annual influenza outbreaks, pandemic influenza, SARS, bioweapons attacks; 4) adherence monitoring for selected infection control practices; 5) oversight of risk assessment and implementation of prevention measures associated with construction and renovation; 6) prevention of transmission of MDROs; 7) evaluation of new medical products that could be associated with increased infection risk. e.g.,intravenous infusion materials; 9) communication with the public, facility staff, and state and local health departments concerning infection control-related issues; and 10) participation in local and multi-center research projects [434, 549, 552, 558, 573, 574].

None of the CBIC job analyses addressed specific staffing requirements for the identified tasks, although the surveys did include information about hours worked; the 2001 survey included the number of ICPs assigned to the responding facilities [558]. There is agreement in the literature that 1 ICP per 250 acute care beds is no longer adequate to meet current infection control needs; a Delphi project that assessed staffing needs of infection control programs in the 21st century concluded that a ratio of 0.8 to 1.0 ICP per 100 occupied acute care beds is an appropriate level of staffing [552]. A survey of participants in the National

Nosocomial Infections Surveillance (NNIS) system found the average daily census per ICP was 115 [316]. Results of other studies have been similar: 3 per 500 beds for large acute care hospitals, 1 per 150-250 beds in long term care facilities, and 1.56 per 250 in small rural hospitals [573, 575]. The foregoing demonstrates that infection control staffing can no longer be based on patient census alone, but rather must be determined by the scope of the program, characteristics of the patient population, complexity of the healthcare system, tools available to assist personnel to perform essential tasks (e.g., electronic tracking and laboratory support for surveillance), and unique or urgent needs of the institution and community [552]. Furthermore, appropriate training is required to optimize the quality of work performed [558, 572, 576].

II.A.1.a.i. Infection Control Nurse Liaison Designating a bedside nurse on a patient care unit as an infection control liaison or "link nurse" is reported to be an effective adjunct to enhance infection control at the unit level [577-582]. Such individuals receive training in basic infection control and have frequent communication with the ICPs, but maintain their primary role as bedside caregiver on their units. The infection control nurse liaison increases the awareness of infection control at the unit level. He or she is especially effective in implementation of new policies or control interventions because of the rapport with individuals on the unit, an understanding of unit-specific challenges, and ability to promote strategies that are most likely to be successful in that unit. This position is an adjunct to, not a replacement for, fully trained ICPs. Furthermore, the infection control liaison nurses should not be counted when considering ICP staffing.

II.A.1.b. Bedside nurse staffing There is increasing evidence that the level of bedside nurse-staffing influences the quality of patient care [583, 584]. If there are adequate nursing staff, it is more likely that infection control practices, including hand hygiene and Standard and Transmission-Based Precautions, will be given appropriate attention and applied correctly and consistently [552]. A national multicenter study reported strong and consistent inverse relationships between nurse staffing and five adverse outcomes in medical patients, two of which were HAIs: urinary tract infections and pneumonia [583]. The association of nursing staff shortages with increased rates of HAIs has been demonstrated in several outbreaks in hospitals and long term care settings, and with increased transmission of hepatitis C virus in dialysis units [22, 418, 551, 585-597]. In most cases, when staffing improved as part of a comprehensive control intervention, the outbreak ended or the HAI rate declined. In two studies [590, 596], the composition of the nursing staff ("pool" or "float" vs. regular staff nurses) influenced the rate of primary bloodstream infections, with an increased infection rate occurring when the proportion of regular nurses decreased and pool nurses increased.

II.A.1.c. Clinical microbiology laboratory support The critical role of the clinical microbiology laboratory in infection control and healthcare epidemiology is described well [553, 554, 598-600] and is supported by the Infectious Disease Society

of America policy statement on consolidation of clinical microbiology laboratories published in 2001 [553]. The clinical microbiology laboratory contributes to preventing transmission of infectious diseases in healthcare settings by promptly detecting and reporting epidemiologically important organisms, identifying emerging patterns of antimicrobial resistance, and assisting in assessment of the effectiveness of recommended precautions to limit transmission during outbreaks [598]. Outbreaks of infections may be recognized first by laboratorians [162]. Healthcare organizations need to ensure the availability of the recommended scope and quality of laboratory services, a sufficient number of appropriately trained laboratory staff members, and systems to promptly communicate epidemiologically important results to those who will take action (e.g., providers of clinical care, infection control staff, healthcare epidemiologists, and infectious disease consultants) [601]. As concerns about emerging pathogens and bioterrorism grow, the role of the clinical microbiology laboratory takes on even greater importance. For healthcare organizations that outsource microbiology laboratory services (e.g., ambulatory care, home care, LTCFs, smaller acute care hospitals), it is important to specify by contract the types of services (e.g., periodic institution-specific aggregate susceptibility reports) required to support infection control.

Several key functions of the clinical microbiology laboratory are relevant to this guideline:

- Antimicrobial susceptibility by testing and interpretation in accordance with current guidelines developed by the National Committee for Clinical Laboratory Standards (NCCLS), known as the Clinical and Laboratory Standards Institute (CLSI) since 2005 [602], for the detection of emerging resistance patterns [603, 604], and for the preparation, analysis, and distribution of periodic cumulative antimicrobial susceptibility summary reports [605-607]. While not required, clinical laboratories ideally should have access to rapid genotypic identification of bacteria and their antibiotic resistance genes [608].
- Performance of surveillance cultures when appropriate (including retention of isolates for analysis) to assess patterns of infection transmission and effectiveness of infection control interventions at the facility or organization. Microbiologists assist in decisions concerning the indications for initiating and discontinuing active surveillance programs and optimize the use of laboratory resources.
- Molecular typing, on-site or outsourced, in order to investigate and control healthcare-associated outbreaks [609].
- Application of rapid diagnostic tests to support clinical decisions involving patient treatment, room selection, and implementation of control measures including barrier precautions and use of vaccine or chemoprophylaxis agents (e.g., influenza [610-612], B. pertussis [613], RSV [614, 615], and enteroviruses [616]). The microbiologist provides guidance to limit rapid testing to clinical situations in which rapid results influence patient

management decisions, as well as providing oversight of point-of-care testing performed by non-laboratory healthcare workers [617].

- Detection and rapid reporting of epidemiologically important organisms, including those that are reportable to public health agencies.
- Implementation of a quality control program that ensures testing services are appropriate for the population served, and stringently evaluated for sensitivity, specificity, applicability, and feasibility.
- Participation in a multidisciplinary team to develop and maintain an effective institutional program for the judicious use of antimicrobial agents [618, 619].

II.A.2. Institutional safety culture and organizational characteristics Safety culture (or safety climate) refers to a work environment where a shared commitment to safety on the part of management and the workforce is understood and followed [557, 620, 621]. The authors of the Institute of Medicine Report, To Err is Human [543], acknowledge that causes of medical error are multifaceted but emphasize repeatedly the pivotal role of system failures and the benefits of a safety culture. A safety culture is created through 1) the actions management takes to improve patient and worker safety; 2) worker participation in safety planning; 3) the availability of appropriate protective equipment; 4) influence of group norms regarding acceptable safety practices; and 5) the organization's socialization process for new personnel. Safety and patient outcomes can be enhanced by improving or creating organizational characteristics within patient care units as demonstrated by studies of surgical ICUs [622, 623]. Each of these factors has a direct bearing on adherence to transmission prevention recommendations [257]. Measurement of an institutional culture of safety is useful for designing improvements in healthcare [624, 625]. Several hospital-based studies have linked measures of safety culture with both employee adherence to safe practices and reduced exposures to blood and body fluids [626-632]. One study of hand hygiene practices concluded that improved adherence requires integration of infection control into the organization's safety culture [561]. Several hospitals that are part of the Veterans Administration Healthcare System have taken specific steps toward improving the safety culture, including error reporting mechanisms, performing root cause analysis on problems identified, providing safety incentives, and employee education. [633-635].

II.A.3. Adherence of healthcare personnel to recommended guidelines Adherence to recommended infection control practices decreases transmission of infectious agents in healthcare settings [116, 562, 636-640]. However, several observational studies have shown limited adherence to recommended practices by healthcare personnel [559, 640-657]. Observed adherence to universal precautions ranged from 43% to 89% [641, 642, 649, 651, 652]. However, the degree of adherence depended frequently on the practice that was assessed and, for glove use, the circumstance in which they were used. Appropriate glove use has ranged from a low of 15% [645] to a high of 82% [650]. However, 92% and 98% adherence with glove use have been reported during arterial blood gas collection and

resuscitation, respectively, procedures where there may be considerable blood contact [643, 656]. Differences in observed adherence have been reported among occupational groups in the same healthcare facility [641] and between experienced and nonexperienced professionals [645]. In surveys of healthcare personnel, self-reported adherence was generally higher than that reported in observational studies. Furthermore, where an observational component was included with a self-reported survey, self-perceived adherence was often greater than observed adherence [657]. Among nurses and physicians, increasing years of experience is a negative predictor of adherence [645, 651]. Education to improve adherence is the primary intervention that has been studied. While positive changes in knowledge and attitude have been demonstrated, [640, 658], there often has been limited or no accompanying change in behavior [642, 644]. Self-reported adherence is higher in groups that have received an educational intervention [630, 659]. Educational interventions that incorporated videotaping and performance feedback were successful in improving adherence during the period of study; the long-term effect of these interventions is not known [654].The use of videotape also served to identify system problems (e.g., communication and access to personal protective equipment) that otherwise may not have been recognized.

Use of engineering controls and facility design concepts for improving adherence is gaining interest. While introduction of automated sinks had a negative impact on consistent adherence to hand washing [660], use of electronic monitoring and voice prompts to remind healthcare workers to perform hand hygiene, and improving accessibility to hand hygiene products, increased adherence and contributed to a decrease in HAIs in one study [661]. More information is needed regarding how technology might improve adherence.

Improving adherence to infection control practices requires a multifaceted approach that incorporates continuous assessment of both the individual and the work environment [559, 561]. Using several behavioral theories, Kretzer and Larson concluded that a single intervention (e.g., a handwashing campaign or putting up new posters about transmission precautions) would likely be ineffective in improving healthcare personnel adherence [662]. Improvement requires that the organizational leadership make prevention an institutional priority and integrate infection control practices into the organization's safety culture [561]. A recent review of the literature concluded that variations in organizational factors (e.g., safety climate, policies and procedures, education and training) and individual factors (e.g., knowledge, perceptions of risk, past experience) were determinants of adherence to infection control guidelines for protection against SARS and other respiratory pathogens [257].

II.B. Surveillance for healthcare-associated infections (HAIs)

Surveillance is an essential tool for case-finding of single patients or clusters of patients who are infected or colonized with epidemiologically important organisms (e.g., susceptible bacteria such as *S. aureus, S. pyogenes* [Group A streptococcus] or Enterobacter-Klebsiella spp; MRSA, VRE, and other MDROs; *C. difficile*; RSV; influenza virus) for which transmission-based precautions may

be required. Surveillance is defined as the ongoing, systematic collection, analysis, interpretation, and dissemination of data regarding a health-related event for use in public health action to reduce morbidity and mortality and to improve health [663]. The work of Ignaz Semmelweis that described the role of person-to-person transmission in puerperal sepsis is the earliest example of the use of surveillance data to reduce transmission of infectious agents [664]. Surveillance of both process measures and the infection rates to which they are linked are important for evaluating the effectiveness of infection prevention efforts and identifying indications for change [555, 665-668].

The Study on the Efficacy of Nosocomial Infection Control (SENIC) found that different combinations of infection control practices resulted in reduced rates of nosocomial surgical site infections, pneumonia, urinary tract infections, and bacteremia in acute care hospitals [566]; however, surveillance was the only component essential for reducing all four types of HAIs. Although a similar study has not been conducted in other healthcare settings, a role for surveillance and the need for novel strategies have been described in LTCFs [398, 434, 669, 670] and in home care [470-473]. The essential elements of a surveillance system are: 1) standardized definitions; 2) identification of patient populations at risk for infection; 3) statistical analysis (e.g. risk-adjustment, calculation of rates using appropriate denominators, trend analysis using methods such as statistical process control charts); and 4) feedback of results to the primary caregivers [671-676]. Data gathered through surveillance of high-risk populations, device use, procedures, and/or facility locations (e.g., ICUs) are useful for detecting transmission trends [671-673]. Identification of clusters of infections should be followed by a systematic epidemiologic investigation to determine commonalities in persons, places, and time; and guide implementation of interventions and evaluation of the effectiveness of those interventions.

Targeted surveillance based on the highest risk areas or patients has been preferred over facility-wide surveillance for the most effective use of resources [673, 676]. However, surveillance for certain epidemiologically important organisms may need to be facility-wide. Surveillance methods will continue to evolve as healthcare delivery systems change [392, 677] and user-friendly electronic tools become more widely available for electronic tracking and trend analysis [674, 678, 679]. Individuals with experience in healthcare epidemiology and infection control should be involved in selecting software packages for data aggregation and analysis to assure that the need for efficient and accurate HAI surveillance will be met. Effective surveillance is increasingly important as legislation requiring public reporting of HAI rates is passed and states work to develop effective systems to support such legislation [680].

II.C. Education of HCWs, patients, and families

Education and training of healthcare personnel are a prerequisite for ensuring that policies and procedures for Standard and Transmission-Based Precautions are understood and practiced. Understanding the scientific rationale for the

precautions will allow HCWs to apply procedures correctly, as well as safely modify precautions based on changing requirements, resources, or healthcare settings [14, 655, 681-688]. In one study, the likelihood of HCWs developing SARS was strongly associated with less than 2 hours of infection control training and lack of understanding of infection control procedures [689]. Education about the important role of vaccines (e.g., influenza, measles, varicella, pertussis, pneumococcal) in protecting healthcare personnel, their patients, and family members can help improve vaccination rates [690-693].

Education on the principles and practices for preventing transmission of infectious agents should begin during training in the health professions and be provided to anyone who has an opportunity for contact with patients or medical equipment (e.g., nursing and medical staff; therapists and technicians, including respiratory, physical, occupational, radiology, and cardiology personnel; phlebotomists; housekeeping and maintenance staff; and students). In healthcare facilities, education and training on Standard and Transmission-Based Precautions are typically provided at the time of orientation and should be repeated as necessary to maintain competency; updated education and training are necessary when policies and procedures are revised or when there is a special circumstance, such as an outbreak that requires modification of current practice or adoption of new recommendations. Education and training materials and methods appropriate to the HCW's level of responsibility, individual learning habits, and language needs, can improve the learning experience [658, 694-702].

Education programs for healthcare personnel have been associated with sustained improvement in adherence to best practices and a related decrease in device-associated HAIs in teaching and non-teaching settings [639, 703] and in medical and surgical ICUs {Coopersmith, 2002 #2149; Babcock, 2004 #2126; Berenholtz, 2004 #2289; www.ihi.org/IHI/Programs/Campaign, #2563}. Several studies have shown that, in addition to targeted education to improve specific practices, periodic assessment and feedback of the HCWs knowledge,and adherence to recommended practices are necessary to achieve the desired changes and to identify continuing education needs [562, 704-708]. Effectiveness of this approach for isolation practices has been demonstrated for control of RSV [116, 684].

Patients, family members, and visitors can be partners in preventing transmission of infections in healthcare settings [9, 42, 709-711]. Information about Standard Precautions, especially hand hygiene, Respiratory Hygiene/Cough Etiquette, vaccination (especially against influenza) and other routine infection prevention strategies may be incorporated into patient information materials that are provided upon admission to the healthcare facility. Additional information about Transmission-Based Precautions is best provided at the time they are initiated. Fact sheets, pamphlets, and other printed material may include information on the rationale for the additional precautions, risks to household members, room assignment for Transmission-Based Precautions purposes, explanation about the use of personal protective equipment by HCWs, and directions for use of

such equipment by family members and visitors. Such information may be particularly helpful in the home environment where household members often have primary responsibility for adherence to recommended infection control practices. Healthcare personnel must be available and prepared to explain this material and answer questions as needed.

II.D. Hand hygiene

Hand hygiene has been cited frequently as the single most important practice to reduce the transmission of infectious agents in healthcare settings [559, 712, 713] and is an essential element of Standard Precautions. The term "hand hygiene" includes both handwashing with either plain or antiseptic-containing soap and water, and use of alcohol-based products (gels, rinses, foams) that do not require the use of water. In the absence of visible soiling of hands, approved alcohol-based products for hand disinfection are preferred over antimicrobial or plain soap and water because of their superior microbiocidal activity, reduced drying of the skin, and convenience [559]. Improved hand hygiene practices have been associated with a sustained decrease in the incidence of MRSA and VRE infections primarily in the ICU [561, 562, 714-717]. The scientific rationale, indications, methods, and products for hand hygiene are summarized in other publications [559, 717].

The effectiveness of hand hygiene can be reduced by the type and length of fingernails [559, 718, 719]. Individuals wearing artifical nails have been shown to harbor more pathogenic organisms, especially gram negative bacilli and yeasts, on the nails and in the subungual area than those with native nails [720, 721]. In 2002, CDC/HICPAC recommended (Category IA) that artificial fingernails and extenders not be worn by healthcare personnel who have contact with high-risk patients (e.g., those in ICUs, ORs) due to the association with outbreaks of gram-negative bacillus and candidal infections as confirmed by molecular typing of isolates [30, 31, 559, 722-725].The need to restrict the wearing of artificial fingernails by all healthcare personnel who provide direct patient care or by healthcare personnel who have contact with other high risk groups (e.g., oncology, cystic fibrosis patients), has not been studied, but has been recommended by some experts [20]. At this time such decisions are at the discretion of an individual facility's infection control program. There is less evidence that jewelry affects the quality of hand hygiene. Although hand contamination with potential pathogens is increased with ring-wearing [559, 726], no studies have related this practice to HCW-to-patient transmission of pathogens.

II.E. Personal protective equipment (PPE) for healthcare personnel

PPE refers to a variety of barriers and respirators used alone or in combination to protect mucous membranes, airways, skin, and clothing from contact with infectious agents. The selection of PPE is based on the nature of the patient

interaction and/or the likely mode(s) of transmission. Guidance on the use of PPE is discussed in Part III. A suggested procedure for donning and removing PPE that will prevent skin or clothing contamination is presented in the Figure. Designated containers for used disposable or reusable PPE should be placed in a location that is convenient to the site of removal to facilitate disposal and containment of contaminated materials. Hand hygiene is always the final step after removing and disposing of PPE. The following sections highlight the primary uses and methods for selecting this equipment.

II.E.1. Gloves Gloves are used to prevent contamination of healthcare personnel hands when 1) anticipating direct contact with blood or body fluids, mucous membranes, nonintact skin and other potentially infectious material; 2) having direct contact with patients who are colonized or infected with pathogens transmitted by the contact route e.g., VRE, MRSA, RSV [559, 727, 728]; or 3) handling or touching visibly or potentially contaminated patient care equipment and environmental surfaces [72, 73, 559]. Gloves can protect both patients and healthcare personnel from exposure to infectious material that may be carried on hands [73]. The extent to which gloves will protect healthcare personnel from transmission of bloodborne pathogens (e.g., HIV, HBV, HCV) following a needlestick or other pucture that penetrates the glove barrier has not been determined. Although gloves may reduce the volume of blood on the external surface of a sharp by 46-86% [729], the residual blood in the lumen of a hollowbore needle would not be affected; therefore, the effect on transmission risk is unknown.

Gloves manufactured for healthcare purposes are subject to FDA evaluation and clearance [730]. Nonsterile disposable medical gloves made of a variety of materials (e.g., latex, vinyl, nitrile) are available for routine patient care [731]. The selection of glove type for non-surgical use is based on a number of factors, including the task that is to be performed, anticipated contact with chemicals and chemotherapeutic agents, latex sensitivity, sizing, and facility policies for creating a latex-free environment [17, 732-734]. For contact with blood and body fluids during non-surgical patient care, a single pair of gloves generally provides adequate barrier protection [734]. However, there is considerable variability among gloves; both the quality of the manufacturing process and type of material influence their barrier effectiveness [735]. While there is little difference in the barrier properties of unused intact gloves [736], studies have shown repeatedly that vinyl gloves have higher failure rates than latex or nitrile gloves when tested under simulated and actual clinical conditions [731, 735-738]. For this reason either latex or nitrile gloves are preferable for clinical procedures that require manual dexterity and/or will involve more than brief patient contact. It may be necessary to stock gloves in several sizes. Heavier, reusable utility gloves are indicated for non-patient care activities, such as handling or cleaning contaminated equipment or surfaces [11, 14, 739].

During patient care, transmission of infectious organisms can be reduced by adhering to the principles of working from “clean” to “dirty”, and confining or limiting contamination to surfaces that are directly needed for patient care. It may be necessary to change gloves during the care of a single patient to prevent

cross-contamination of body sites [559, 740]. It also may be necessary to change gloves if the patient interaction also involves touching portable computer keyboards or other mobile equipment that is transported from room to room. Discarding gloves between patients is necessary to prevent transmission of infectious material. Gloves must not be washed for subsequent reuse because microorganisms cannot be removed reliably from glove surfaces and continued glove integrity cannot be ensured. Furthermore, glove reuse has been associated with transmission of MRSA and gram-negative bacilli [741-743].
When gloves are worn in combination with other PPE, they are put on last. Gloves that fit snugly around the wrist are preferred for use with an isolation gown because they will cover the gown cuff and provide a more reliable continuous barrier for the arms, wrists, and hands. Gloves that are removed properly will prevent hand contamination (Figure). Hand hygiene following glove removal further ensures that the hands will not carry potentially infectious material that might have penetrated through unrecognized tears or that could contaminate the hands during glove removal [559, 728, 741].

II.E.2. Isolation gowns Isolation gowns are used as specified by Standard and Transmission-Based Precautions, to protect the HCW's arms and exposed body areas and prevent contamination of clothing with blood, body fluids, and other potentially infectious material [24, 88, 262, 744-746]. The need for and type of isolation gown selected is based on the nature of the patient interaction, including the anticipated degree of contact with infectious material and potential for blood and body fluid penetration of the barrier. The wearing of isolation gowns and other protective apparel is mandated by the OSHA Bloodborne Pathogens Standard [739]. Clinical and laboratory coats or jackets worn over personal clothing for comfort and/or purposes of identity are not considered PPE.
When applying Standard Precautions, an isolation gown is worn only if contact with blood or body fluid is anticipated. However, when Contact Precautions are used (i.e., to prevent transmission of an infectious agent that is not interrupted by Standard Precautions alone and that is associated with environmental contamination), donning of both gown and gloves upon room entry is indicated to address unintentional contact with contaminated environmental surfaces [54, 72, 73, 88]. The routine donning of isolation gowns upon entry into an intensive care unit or other high-risk area does not prevent or influence potential colonization or infection of patients in those areas[365, 747-750].

Isolation gowns are always worn in combination with gloves, and with other PPE when indicated. Gowns are usually the first piece of PPE to be donned. Full coverage of the arms and body front, from neck to the mid-thigh or below will ensure that clothing and exposed upper body areas are protected. Several gown sizes should be available in a healthcare facility to ensure appropriate coverage for staff members. Isolation gowns should be removed before leaving the patient care area to prevent possible contamination of the environment outside the patient's room. Isolation gowns should be removed in a manner that prevents contamination of clothing or skin (Figure). The outer, "contaminated", side of the

gown is turned inward and rolled into a bundle, and then discarded into a designated container for waste or linen to contain contamination.

II.E.3. Face protection: masks, goggles, face shields

II.E.3.a. *Masks* Masks are used for three primary purposes in healthcare settings: 1) placed on healthcare personnel to protect them from contact with infectious material from patients e.g., respiratory secretions and sprays of blood or body fluids, consistent with Standard Precautions and Droplet Precautions; 2) placed on healthcare personnel when engaged in procedures requiring sterile technique to protect patients from exposure to infectious agents carried in a healthcare worker's mouth or nose, and 3) placed on coughing patients to limit potential dissemination of infectious respiratory secretions from the patient to others (i.e., Respiratory Hygiene/Cough Etiquette). Masks may be used in combination with goggles to protect the mouth, nose and eyes, or a face shield may be used instead of a mask and goggles, to provide more complete protection for the face, as discussed below. **Masks should not be confused with particulate respirators that are used to prevent inhalation of small particles that may contain infectious agents transmitted via the airborne route as described below.**

The mucous membranes of the mouth, nose, and eyes are susceptible portals of entry for infectious agents, as can be other skin surfaces if skin integrity is compromised (e.g., by acne, dermatitis) [66, 751-754]. Therefore, use of PPE to protect these body sites is an important component of Standard Precautions. The protective effect of masks for exposed healthcare personnel has been demonstrated [93, 113, 755, 756]. Procedures that generate splashes or sprays of blood, body fluids, secretions, or excretions (e.g., endotracheal suctioning, bronchoscopy, invasive vascular procedures) require either a face shield (disposable or reusable) or mask and goggles [93-95, 96 , 113, 115, 262, 739, 757] .The wearing of masks, eye protection, and face shields in specified circumstances when blood or body fluid exposures are likely to occur is mandated by the OSHA Bloodborne Pathogens Standard [739]. Appropriate PPE should be selected based on the anticipated level of exposure.

Two mask types are available for use in healthcare settings: surgical masks that are cleared by the FDA and required to have fluid-resistant properties, and procedure or isolation masks [758 #2688]. No studies have been published that compare mask types to determine whether one mask type provides better protection than another. Since procedure/isolation masks are not regulated by the FDA, there may be more variability in quality and performance than with surgical masks. Masks come in various shapes (e.g., molded and non-molded), sizes, filtration efficiency, and method of attachment (e.g., ties, elastic, ear loops). Healthcare facilities may find that different types of masks are needed to meet individual healthcare personnel needs.

II.E.3.b. *Goggles, face shields* Guidance on eye protection for infection control has been published [759]. The eye protection chosen for specific work situations (e.g., goggles or face shield) depends upon the circumstances of exposure, other

PPE used, and personal vision needs. Personal eyeglasses and contact lenses are NOT considered adequate eye protection (www.cdc.gov/niosh/topics/eye/eye-infectious.html). NIOSH states that, eye protection must be comfortable, allow for sufficient peripheral vision, and must be adjustable to ensure a secure fit. It may be necessary to provide several different types, styles, and sizes of protective equipment. Indirectly-vented goggles with a manufacturer's anti-fog coating may provide the most reliable practical eye protection from splashes, sprays, and respiratory droplets from multiple angles. Newer styles of goggles may provide better indirect airflow properties to reduce fogging, as well as better peripheral vision and more size options for fitting goggles to different workers. Many styles of goggles fit adequately over prescription glasses with minimal gaps. While effective as eye protection, goggles do not provide splash or spray protection to other parts of the face.

The role of goggles, in addition to a mask, in preventing exposure to infectious agents transmitted via respiratory droplets has been studied only for RSV. Reports published in the mid-1980s demonstrated that eye protection reduced occupational transmission of RSV [760, 761]. Whether this was due to preventing hand-eye contact or respiratory droplet-eye contact has not been determined. However, subsequent studies demonstrated that RSV transmission is effectively prevented by adherence to Standard plus Contact Precations and that for this virus routine use of goggles is not necessary [24, 116, 117, 684, 762]. It is important to remind healthcare personnel that even if Droplet Precautions are not recommended for a specific respiratory tract pathogen, protection for the eyes, nose and mouth by using a mask and goggles, or face shield alone, is necessary when it is likely that there will be a splash or spray of any respiratory secretions or other body fluids as defined in Standard Precautions

Disposable or non-disposable face shields may be used as an alternative to goggles [759]. As compared with goggles, a face shield can provide protection to other facial areas in addition to the eyes. Face shields extending from chin to crown provide better face and eye protection from splashes and sprays; face shields that wrap around the sides may reduce splashes around the edge of the shield.

Removal of a face shield, goggles and mask can be performed safely after gloves have been removed, and hand hygiene performed. The ties, ear pieces and/or headband used to secure the equipment to the head are considered "clean" and therefore safe to touch with bare hands. The front of a mask, goggles and face shield are considered contaminated (Figure).

II.E.4. Respiratory protection The subject of respiratory protection as it applies to preventing transmission of airborne infectious agents, including the need for and frequency of fit-testing is under scientific review and was the subject of a CDC workshop in 2004 [763]. Respiratory protection currently requires the use of a respirator with N95 or higher filtration to prevent inhalation of infectious particles. Information about respirators and respiratory protection programs is summarized

in the *Guideline for Preventing Transmission of Mycobacterium tuberculosis in Health-care Settings, 2005* (CDC.MMWR 2005; 54: RR-17 [12]).
Respiratory protection is broadly regulated by OSHA under the general industry standard for respiratory protection (29CFR1910.134)[764] which requires that U.S. employers in all employment settings implement a program to protect employees from inhalation of toxic materials. OSHA program components include medical clearance to wear a respirator; provision and use of appropriate respirators, including fit-tested NIOSH-certified N95 and higher particulate filtering respirators; education on respirator use and periodic re-evaluation of the respiratory protection program. When selecting particulate respirators, models with inherently good fit characteristics (i.e., those expected to provide protection factors of 10 or more to 95% of wearers) are preferred and could theoretically relieve the need for fit testing [765, 766]. Issues pertaining to respiratory protection remain the subject of ongoing debate. Information on various types of respirators may be found at www.cdc.gov/niosh/npptl/respirators/respsars.html and in published studies [765, 767, 768]. A user-seal check (formerly called a "fit check") should be performed by the wearer of a respirator each time a respirator is donned to minimize air leakage around the facepiece [769]. The optimal frequency of fit-testng has not been determined; re-testing may be indicated if there is a change in facial features of the wearer, onset of a medical condition that would affect respiratory function in the wearer, or a change in the model or size of the initially assigned respirator [12].
Respiratory protection was first recommended for protection of preventing U.S. healthcare personnel from exposure to *M. tuberculosis* in 1989. That recommendation has been maintained in two successive revisions of the Guidelines for Prevention of Transmission of Tuberculosis in Hospitals and other Healthcare Settings [12, 126]. The incremental benefit from respirator use, in addition to administrative and engineering controls (i.e., AIIRs, early recognition of patients likely to have tuberculosis and prompt placement in an AIIR, and maintenance of a patient with suspected tuberculosis in an AIIR until no longer infectious), for preventing transmission of airborne infectious agents (e.g., *M. tuberculosis)* is undetermined. Although some studies have demonstrated effective prevention of *M. tuberculosis* transmission in hospitals where surgical masks, instead of respirators, were used in conjunction with other administrative and engineering controls [637, 770, 771], CDC currently recommends N95 or higher level respirators for personnel exposed to patients with suspected or confirmed tuberculosis. Currently this is also true for other diseases that could be transmitted through the airborne route, including SARS [262] and smallpox [108, 129, 772], until inhalational transmission is better defined or healthcare-specific protective equipment more suitable for for preventing infection are developed. Respirators are also currently recommended to be worn during the performance of aerosol-generating procedures (e.g., intubation, bronchoscopy, suctioning) on patients withSARS Co-V infection, avian influenza and pandemic influenza (See Appendix A).
Although Airborne Precautions are recommended for preventing airborne transmission of measles and varicella-zoster viruses, there are no data upon

which to base a recommendation for respiratory protection to protect susceptible personnel against these two infections; transmission of varicella-zoster virus has been prevented among pediatric patients using negative pressure isolation alone [773]. Whether respiratory protection (i.e., wearing a particulate respirator) would enhance protection from these viruses has not been studied. Since the majority of healthcare personnel have natural or acquired immunity to these viruses, only immune personnel generally care for patients with these infections [774-777]. Although there is no evidence to suggest that masks are not adequate to protect healthcare personnel in these settings, for purposes of consistency and simplicity, or because of difficulties in ascertaining immunity, some facilities may require the use of respirators for entry into all AIIRs, regardless of the specific infectious agent.

Procedures for safe removal of respirators are provided (Figure). In some healthcare settings, particulate respirators used to provide care for patients *with M. tuberculosis* are reused by the same HCW. This is an acceptable practice providing the respirator is not damaged or soiled, the fit is not compromised by change in shape, and the respirator has not been contaminated with blood or body fluids. There are no data on which to base a recommendation for the length of time a respirator may be reused.

II.F. Safe work practices to prevent HCW exposure to bloodborne pathogens

II.F.1. Prevention of needlesticks and other sharps-related injuries Injuries due to needles and other sharps have been associated with transmission of HBV, HCV and HIV to healthcare personnel [778, 779]. The prevention of sharps injuries has always been an essential element of Universal and now Standard Precautions [1, 780]. These include measures to handle needles and other sharp devices in a manner that will prevent injury to the user and to others who may encounter the device during or after a procedure. These measures apply to routine patient care and do not address the prevention of sharps injuries and other blood exposures during surgical and other invasive procedures that are addressed elsewhere [781-785].

Since 1991, when OSHA first issued its Bloodborne Pathogens Standard to protect healthcare personnel from blood exposure, the focus of regulatory and legislative activity has been on implementing a hierarchy of control measures. This has included focusing attention on removing sharps hazards through the development and use of engineering controls. The federal Needlestick Safety and Prevention Act signed into law in November, 2000 authorized OSHA's revision of its Bloodborne Pathogens Standard to more explicitly require the use of safety-engineered sharp devices [786]. CDC has provided guidance on sharps injury prevention [787, 788], including for the design, implementation and evaluation of a comprehensive sharps injury prevention program [789].

II.F.2. Prevention of mucous membrane contact Exposure of mucous membranes of the eyes, nose and mouth to blood and body fluids has been associated with the transmission of bloodborne viruses and other infectious agents to healthcare personnel [66, 752, 754, 779]. The prevention of mucous membrane exposures has always been an element of Universal and now Standard Precautions for routine patient care [1, 753] and is subject to OSHA bloodborne pathogen regulations. Safe work practices, in addition to wearing PPE, are used to protect mucous membranes and non-intact skin from contact with potentially infectious material. These include keeping gloved and ungloved hands that are contaminated from touching the mouth, nose, eyes, or face; and positioning patients to direct sprays and splatter away from the face of the caregiver. Careful placement of PPE before patient contact will help avoid the need to make PPE adjustments and possible face or mucous membrane contamination during use.
In areas where the need for resuscitation is unpredictable, mouthpieces, pocket resuscitation masks with one-way valves, and other ventilation devices provide an alternative to mouth-to-mouth resuscitation, preventing exposure of the caregiver's nose and mouth to oral and respiratory fluids during the procedure.

II.F.2.a. Precautions during aerosol-generating procedures The performance of procedures that can generate small particle aerosols (aerosol-generating procedures), such as bronchoscopy, endotracheal intubation, and open suctioning of the respiratory tract, have been associated with transmission of infectious agents to healthcare personnel, including *M. tuberculosis* [790], SARS-CoV [93, 94, 98] and *N. meningitidis* [95]. Protection of the eyes, nose and mouth, in addition to gown and gloves, is recommended during performance of these procedures in accordance with Standard Precautions. Use of a particulate respirator is recommended during aerosol-generating procedures when the aerosol is likely to contain *M. tuberculosis*, SARS-CoV, or avian or pandemic influenza viruses.

II.G. Patient placement

II.G.1. Hospitals and long-term care settings Options for patient placement include single patient rooms, two patient rooms, and multi-bed wards. Of these, single patient rooms are prefered when there is a concern about transmission of an infectious agent. Although some studies have failed to demonstrate the efficacy of single patient rooms to prevent HAIs [791], other published studies, including one commissioned by the American Institute of Architects and the Facility Guidelines Institute, have documented a beneficial relationship between private rooms and reduction in infectious and noninfectious adverse patient outcomes [792, 793]. The AIA notes that private rooms are the trend in hospital planning and design. However, most hospitals and long-term care facilities have multi-bed rooms and must consider many competing priorities when determining the appropriate room placement for patients (e.g., reason for admission; patient characteristics, such as age, gender, mental status; staffing needs; family

requests; psychosocial factors; reimbursement concerns). In the absence of obvious infectious diseases that require specified airborne infection isolation rooms (e.g., tuberculosis, SARS, chickenpox), the risk of transmission of infectious agents is not always considered when making placement decisions**.** When there are only a limited number of single-patient rooms, it is prudent to prioritize them for those patients who have conditions that facilitate transmission of infectious material to other patients (e.g., draining wounds, stool incontinence, uncontained secretions) and for those who are at increased risk of acquisition and adverse outcomes resulting from HAI (e.g., immunosuppression, open wounds, indwelling catheters, anticipated prolonged length of stay, total dependence on HCWs for activities of daily living) [15, 24, 43, 430, 794, 795].

Single-patient rooms are always indicated for patients placed on Airborne Precautions and in a Protective Environment and are preferred for patients who require Contact or Droplet Precautions [23, 24, 410, 435, 796, 797]. During a suspected or proven outbreak caused by a pathogen whose reservoir is the gastrointestinal tract, use of single patient rooms with private bathrooms limits opportunities for transmission, especially when the colonized or infected patient has poor personal hygiene habits, fecal incontinence, or cannot be expected to assist in maintaining procedures that prevent transmission of microorganisms (e.g., infants, children, and patients with altered mental status or developmental delay). In the absence of continued transmission, it is not necessary to provide a private bathroom for patients colonized or infected with enteric pathogens as long as personal hygiene practices and Standard Precautions, especially hand hygiene and appropriate environmental cleaning, are maintained. Assignment of a dedicated commode to a patient,and cleaning and disinfecting fixtures and equipment that may have fecal contamination (e.g., bathrooms, commodes [798], scales used for weighing diapers) and the adjacent surfaces with appropriate agents may be especially important when a single-patient room can not be used since environmental contamination with intestinal tract pathogens is likely from both continent and incontinent patients [54, 799]. Results of several studies to determine the benefit of a single-patient room to prevent transmission of *Clostridium difficile* are inconclusive [167, 800-802]. Some studies have shown that being in the same room with a colonized or infected patient is not necessarily a risk factor for transmission [791, 803-805]. However, for children, the risk of healthcare-associated diarrhea is increased with the increased number of patients per room [806]. Thus, patient factors are important determinants of infection transmission risks, and the need for a single-patient room and/or private bathroom for any patient is best determined on a case-by-case basis.

Cohorting is the practice of grouping together patients who are colonized or infected with the same organism to confine their care to one area and prevent contact with other patients. Cohorts are created based on clinical diagnosis, microbiologic confirmation when available, epidemiology, and mode of transmission of the infectious agent. It is generally preferred not to place severely immunosuppressed patients in rooms with other patients. Cohorting has been used extensively for managing outbreaks of MDROs including MRSA [22, 807], VRE [638, 808, 809], MDR-ESBLs [810]; *Pseudomonas aeruginosa* [29]; methicillin-susceptible

Staphylococcus aureus [811]; RSV [812, 813]; adenovirus keratoconjunctivitis [814]; rotavirus [815]; and SARS [816]. Modeling studies provide additional support for cohorting patients to control outbreaks Talon [817-819]. However, cohorting often is implemented only after routine infection control measures have failed to control an outbreak.
Assigning or cohorting healthcare personnel to care only for patients infected or colonized with a single target pathogen limits further transmission of the target pathogen to uninfected patients [740, 819] but is difficult to achieve in the face of current staffing shortages in hospitals [583] and residential healthcare sites [820-822]. However, when continued transmission is occurring after implementing routine infection control measures and creating patient cohorts, cohorting of healthcare personnel may be beneficial.

During the seasons when RSV, human metapneumovirus [823], parainfluenza, influenza, other respiratory viruses [824], and rotavirus are circulating in the community, cohorting based on the presenting clinical syndrome is often a priority in facilities that care for infants and young children [825]. For example, during the respiratory virus season, infants may be cohorted based soley on the clinical diagnosis of bronchiolitis due to the logistical difficulties and costs associated with requiring microbiologic confirmation prior to room placement, and the predominance of RSV during most of the season. However, when available, single patient rooms are always preferred since a common clinical presentation (e.g., bronchiolitis), can be caused by more than one infectious agent [823, 824, 826]. Furthermore, the inability of infants and children to contain body fluids, and the close physical contact that occurs during their care, increases infection transmission risks for patients and personnel in this setting [24, 795].

II.G.2. Ambulatory settings Patients actively infected with or incubating transmissible infectious diseases are seen frequently in ambulatory settings (e.g., outpatient clinics, physicians' offices, emergency departments) and potentially expose healthcare personnel and other patients, family members and visitors [21, 34, 127, 135, 142, 827]. In response to the global outbreak of SARS in 2003 and in preparation for pandemic influenza, healthcare providers working in outpatient settings are urged to implement source containment measures (e.g., asking couging patients to wear a surgical mask or cover their coughs with tissues) to prevent transmission of respiratory infections, beginning at the point of initial patient encounter [9, 262, 828] as described below in section III.A.1.a. Signs can be posted at the entrance to facilities or at the reception or registration desk requesting that the patient or individuals accompanying the patient promptly inform the receptionist if there are symptoms of a respiratory infection (e.g., cough, flu-like illness, increased production of respiratory secretions). The presence of diarrhea, skin rash, or known or suspected exposure to a transmissible disease (e.g., measles, pertussis, chickenpox, tuberculosis) also could be added. Placement of potentially infectious patients without delay in an examination room limits the number of exposed individuals, e.g., in the common waiting area.

In waiting areas, maintaining a distance between symptomatic and non-symptomatic patients (e.g., >3 feet), in addition to source control measures, may limit exposures. However, infections transmitted via the airborne route (e.g., *M tuberculosis*, measles, chickenpox) require additional precautions [12, 125, 829]. Patients suspected of having such an infection can wear a surgical mask for source containment, if tolerated, and should be placed in an examination room, preferably an AIIR, as soon as possible. If this is not possible, having the patient wear a mask and segregate him/herself from other patients in the waiting area will reduce opportunities to expose others. Since the person(s) accompanying the patient also may be infectious, application of the same infection control precautions may need to be extended to these persons if they are symptomatic [21, 252, 830]. For example, family members accompanying children admitted with suspected *M. tuberculosis* have been found to have unsuspected pulmonary tuberculosis with cavitary lesions, even when asymptomatic [42, 831].
Patients with underlying conditions that increase their susceptibility to infection (e.g., those who are immunocompromised [43, 44] or have cystic fibrosis [20]) require special efforts to protect them from exposures to infected patients in common waiting areas. By informing the receptionist of their infection risk upon arrival, appropriate steps may be taken to further protect them from infection. In some cystic fibrosis clinics, in order to avoid exposure to other patients who could be colonized with *B. cepacia*, patients have been given beepers upon registration so that they may leave the area and receive notification to return when an examination room becomes available [832].

II.G.3. Home care In home care, the patient placement concerns focus on protecting others in the home from exposure to an infectious household member. For individuals who are especially vulnerable to adverse outcomes associated with certain infections, it may be beneficial to either remove them from the home or segregate them within the home. Persons who are not part of the household may need to be prohibited from visiting during the period of infectivity. For example, if a patient with pulmonary tuberculosis is contagious and being cared for at home, very young children (<4 years of age) [833] and immunocompromised persons who have not yet been infected should be removed or excluded from the household. During the SARS outbreak of 2003, segregation of infected persons during the communicable phase of the illness was beneficial in preventing household transmission [249, 834].

II.H. Transport of patients

Several principles are used to guide transport of patients requiring Transmission-Based Precautions. In the inpatient and residential settings these include 1) limiting transport of such patients to essential purposes, such as diagnostic and therapeutic procedures that cannot be performed in the patient's room; 2) when transport is necessary, using appropriate barriers on the patient (e.g., mask, gown, wrapping in sheets or use of impervious dressings to cover the affected area(s) when infectious skin lesions or drainage are present, consistent with the route and risk of transmission; 3) notifying healthcare personnel in the receiving

area of the impending arrival of the patient and of the precautions necessary to prevent transmission; and 4) for patients being transported outside the facility, informing the receiving facility and the medi-van or emergency vehicle personnel in advance about the type of Transmission-Based Precautions being used. For tuberculosis, additional precautions may be needed in a small shared air space such as in an ambulance [12].

II.I. Environmental measures

Cleaning and disinfecting non-critical surfaces in patient-care areas are part of Standard Precautions. In general, these procedures do not need to be changed for patients on Transmission-Based Precautions. The cleaning and disinfection of all patient-care areas is important for frequently touched surfaces, especially those closest to the patient, that are most likely to be contaminated (e.g., bedrails, bedside tables, commodes, doorknobs, sinks, surfaces and equipment in close proximity to the patient) [11, 72, 73, 835]. The frequency or intensity of cleaning may need to change based on the patient's level of hygiene and the degree of environmental contamination and for certain for infectious agents whose reservoir is the intestinal tract [54]. This may be especially true in LTCFs and pediatric facilities where patients with stool and urine incontinence are encountered more frequently. Also, increased frequency of cleaning may be needed in a Protective Environment to minimize dust accumulation [11]. Special recommendations for cleaning and disinfecting environmental surfaces in dialysis centers have been published [18]. In all healthcare settings, administrative, staffing and scheduling activities should prioritize the proper cleaning and disinfection of surfaces that could be implicated in transmission. During a suspected or proven outbreak where an environmental reservoir is suspected, routine cleaning procedures should be reviewed, and the need for additional trained cleaning staff should be assessed. Adherence should be monitored and reinforced to promote consistent and correct cleaning is performed.

EPA-registered disinfectants or detergents/disinfectants that best meet the overall needs of the healthcare facility for routine cleaning and disinfection should be selected [11, 836]. In general, use of the existing facility detergent/disinfectant according to the manufacturer's recommendations for amount, dilution, and contact time is sufficient to remove pathogens from surfaces of rooms where colonized or infected individuals were housed. This includes those pathogens that are resistant to multiple classes of antimicrobial agents (e.g., *C. difficile*, VRE, MRSA, MDR-GNB [11, 24, 88, 435, 746, 796, 837]). Most often, environmental reservoirs of pathogens during outbreaks are related to a failure to follow recommended procedures for cleaning and disinfection rather than the specific cleaning and disinfectant agents used[838-841].

Certain pathogens (e.g., rotavirus, noroviruses, *C. difficile*) may be resistant to some routinely used hospital disinfectants [275, 292, 842-847].The role of specific disinfectants in limiting transmission of rotavirus has been demonstrated experimentally [842]. Also, since *C. difficile* may display increased levels of spore production when exposed to non-chlorine-based cleaning agents, and the spores are more resistant than vegetative cells to commonly used surface disinfectants,

some investigators have recommended the use of a 1:10 dilution of 5.25% sodium hypochlorite (household bleach) and water for routine environmental disinfection of rooms of patients with *C. difficile* when there is continued transmission [844, 848]. In one study, the use of a hypochlorite solution was associated with a decrease in rates of C. difficile infections [847]. The need to change disinfectants based on the presence of these organisms can be determined in consultation with the infection control committee [11, 847, 848]. Detailed recommendations for disinfection and sterilization of surfaces and medical equipment that have been in contact with prion-containing tissue or high risk body fluids, and for cleaning of blood and body substance spills, are available in the Guidelines for Environmental Infection Control in Health-Care Facilities [11] and in the Guideline for Disinfection and Sterilization [848].

II.J. Patient care equipment and instruments/devices

Medical equipment and instruments/devices must be cleaned and maintained according to the manufacturers' instructions to prevent patient-to-patient transmission of infectious agents [86, 87, 325, 849]. Cleaning to remove organic material must always precede high level disinfection and sterilization of critical and semi-critical instruments and devices because residual proteinacous material reduces the effectiveness of the disinfection and sterilization processes [836, 848]. Noncritical equipment, such as commodes, intravenous pumps, and ventilators, must be thoroughly cleaned and disinfected before use on another patient. All such equipment and devices should be handled in a manner that will prevent HCW and environmental contact with potentially infectious material. It is important to include computers and personal digital assistants (PDAs) used in patient care in policies for cleaning and disinfection of non-critical items. The literature on contamination of computers with pathogens has been summarized [850] and two reports have linked computer contamination to colonization and infections in patients [851, 852]. Although keyboard covers and washable keyboards that can be easily disinfected are in use, the infection control benefit of those items and optimal management have not been determined.

In all healthcare settings, providing patients who are on Transmission-Based Precautions with dedicated noncritical medical equipment (e.g., stethoscope, blood pressure cuff, electronic thermometer) has been beneficial for preventing transmission [74, 89, 740, 853, 854]. When this is not possible, disinfection after use is recommended. Consult other guidelines for detailed guidance in developing specific protocols for cleaning and reprocessing medical equipment and patient care items in both routine and special circumstances [11, 14, 18, 20, 740, 836, 848].
In home care, it is preferable to remove visible blood or body fluids from durable medical equipment before it leaves the home. Equipment can be cleaned on-site using a detergent/disinfectant and, when possible, should be placed in a single plastic bag for transport to the reprocessing location [20, 739].

II.K. Textiles and laundry

Soiled textiles, including bedding, towels, and patient or resident clothing may be contaminated with pathogenic microorganisms. However, the risk of disease

transmission is negligible if they are handled, transported, and laundered in a safe manner [11, 855, 856]. Key principles for handling soiled laundry are 1) not shaking the items or handling them in any way that may aerosolize infectious agents; 2) avoiding contact of one's body and personal clothing with the soiled items being handled; and 3) containing soiled items in a laundry bag or designated bin. When laundry chutes are used, they must be maintained to minimize dispersion of aerosols from contaminated items [11]. The methods for handling, transporting, and laundering soiled textiles are determined by organizational policy and any applicable regulations [739]; guidance is provided in the Guidelines for Environmental Infection Control [11]. Rather than rigid rules and regulations, hygienic and common sense storage and processing of clean textiles is recommended [11, 857]. When laundering occurs outside of a healthcare facility, the clean items must be packaged or completely covered and placed in an enclosed space during transport to prevent contamination with outside air or construction dust that could contain infectious fungal spores that are a risk for immunocompromised patients [11].

Institutions are required to launder garments used as personal protective equipment and uniforms visibly soiled with blood or infective material [739]. There are few data to determine the safety of home laundering of HCW uniforms, but no increase in infection rates was observed in the one published study [858] and no pathogens were recovered from home- or hospital-laundered scrubs in another study [859]. In the home, textiles and laundry from patients with potentially transmissible infectious pathogens do not require special handling or separate laundering, and may be washed with warm water and detergent [11, 858, 859].

II.L. Solid waste

The management of solid waste emanating from the healthcare environment is subject to federal and state regulations for medical and non-medical waste [860, 861]. No additional precautions are needed for non-medical solid waste that is being removed from rooms of patients on Transmission-Based Precautions. Solid waste may be contained in a single bag (as compared to using two bags) of sufficient strength. [862].

II.M. Dishware and eating utensils

The combination of hot water and detergents used in dishwashers is sufficient to decontaminate dishware and eating utensils. Therefore, no special precautions are needed for dishware (e.g., dishes, glasses, cups) or eating utensils; reusable dishware and utensils may be used for patients requiring Transmission-Based Precautions. In the home and other communal settings, eating utensils and drinking vessels that are being used should not be shared, consistent with principles of good personal hygiene and for the purpose of preventing transmission of respiratory viruses, *Herpes simplex* virus, and infectious agents that infect the gastrointestinal tract and are transmitted by the fecal/oral route (e.g., hepatitis A virus, noroviruses). If adequate resources for cleaning utensils and dishes are not available, disposable products may be used.

II.N. Adjunctive measures

Important adjunctive measures that are not considered primary components of programs to prevent transmission of infectious agents, but improve the effectiveness of such programs, include 1) antimicrobial management programs; 2) postexposure chemoprophylaxis with antiviral or antibacterial agents; 3) vaccines used both for pre and postexposure prevention; and 4) screening and restricting visitors with signs of transmissible infections. Detailed discussion of judicious use of antimicrobial agents is beyond the scope of this document; however the topic is addressed in the MDRO section (Management of Multidrug-Resistant Organisms in Healthcare Settings 2006. www.cdc.gov/ncidod/dhqp/pdf/ar/mdroGuideline2006.pdf).

II.N.1. Chemoprophylaxis Antimicrobial agents and topical antiseptics may be used to prevent infection and potential outbreaks of selected agents. Infections for which postexposure chemoprophylaxis is recommended under defined conditions include *B. pertussis* [17, 863], *N. meningitidis* [864], *B. anthracis* after environmental exposure to aeosolizable material[865], influenza virus[611], HIV [866], and group A streptococcus [160]. Orally administered antimicrobials may also be used under defined circumstances for MRSA decolonization of patients or healthcare personnel [867].

Another form of chemoprophylaxis is the use of topical antiseptic agents. For example, triple dye is used routinely on the umbilical cords of term newborns to reduce the risk of colonization, skin infections, and omphalitis caused by *S. aureus*, including MRSA, and group A streptococcus [868, 869]. Extension of the use of triple dye to low birth weight infants in the NICU was one component of a program that controlled one longstanding MRSA outbreak [22]. Topical antiseptics are also used for decolonization of healthcare personnel or selected patients colonized with MRSA, using mupirocin as discussed in the MDRO guideline[870, 867, 871-873].

II.N.2. Immunoprophylaxis Certain immunizations recommended for susceptible healthcare personnel have decreased the risk of infection and the potential for transmission in healthcare facilities [17, 874]. The OSHA mandate that requires employers to offer hepatitis B vaccination to HCWs played a substantial role in the sharp decline in incidence of occupational HBV infection [778, 875]. The use of varicella vaccine in healthcare personnel has decreased the need to place susceptible HCWs on administrative leave following exposure to patients with varicella [775]. Also, reports of healthcare-associated transmission of rubella in obstetrical clinics [33, 876] and measles in acute care settings [34] demonstrate the importance of immunization of susceptible healthcare personnel against childhood diseases. Many states have requirements for HCW vaccination for measles and rubella in the absence of evidence of immunity. Annual influenza vaccine campaigns targeted to patients and healthcare personnel in LTCFs and acute-care settings have been instrumental in preventing or limiting institutional

outbreaks and increasing attention is being directed toward improving influenza vaccination rates in healthcare personnel [35 , 611, 690, 877, 878 , 879].
Transmission of *B. pertussis* in healthcare facilities has been associated with large and costly outbreaks that include both healthcare personnel and patients [17, 36, 41, 100, 683, 827, 880, 881]. HCWs who have close contact with infants with pertussis are at particularly high risk because of waning immunity and, until 2005, the absence of a vaccine that could be used in adults. However, two acellular pertussis vaccines were licensed in the United States in 2005, one for use in individuals aged 11-18 and one for use in ages 10-64 years [882]. Provisional ACIP recommendations at the time of publication of this document include adolescents and adults, especially those with contact with infants < 12 months of age and healthcare personnel with direct patient contact [883 884].
Immunization of children and adults will help prevent the introduction of vaccine-preventable diseases into healthcare settings. The recommended immunization schedule for children is published annually in the January issues of the *Morbidity Mortality Weekly Report* with interim updates as needed [885, 886]. An adult immunization schedule also is available for healthy adults and those with special immunization needs due to high risk medical conditions [887].
Some vaccines are also used for postexposure prophylaxis of susceptible individuals, including varicella [888], influenza [611], hepatitis B [778], and smallpox [225] vaccines [17, 874]. In the future, administration of a newly developed *S. aureus* conjugate vaccine (still under investigation) to selected patients may provide a novel method of preventing healthcare-associated *S. aureus,* including MRSA*,* infections in high-risk groups (e.g., hemodialysis patients and candidates for selected surgical procedures) [889, 890].
Immune globulin preparations also are used for postexposure prophylaxis of certain infectious agents under specified circumstances (e.g., varicella-zoster virus [VZIG], hepatitis B virus [HBIG], rabies [RIG], measles and hepatitis A virus [IG] [17, 833, 874]). The RSV monoclonal antibody preparation, Palivizumab, may have contributed to controlling a nosocomial outbreak of RSV in one NICU , but there is insufficient evidence to support a routine recommendation for its use in this setting [891].

II.N. 3. Management of visitors

II.N.3.a. Visitors as sources of infection Visitors have been identified as the source of several types of HAIs (e.g., pertussis [40, 41], *M. tuberculosis* [42, 892], influenza, and other respiratory viruses [24, 43, 44, 373] and SARS [21, 252-254]). However, effective methods for visitor screening in healthcare settings have not been studied. Visitor screening is especially important during community outbreaks of infectious diseases and for high risk patient units. Sibling visits are often encouraged in birthing centers, post partum rooms and in pediatric inpatient units, ICUs, and in residential settings for children; in hospital settings, a child visitor should visit only his or her own sibling. Screening of visiting siblings and other children before they are allowed into clinical areas is necessary to prevent the introduction of childhood illnesses and common respiratory infections.

Screening may be passive through the use of signs to alert family members and visitors with signs and symptoms of communicable diseases not to enter clinical areas. More active screening may include the completion of a screening tool or questionnaire which elicits information related to recent exposures or current symptoms. That information is reviewed by the facility staff and the visitor is either permitted to visit or is excluded [833].
Family and household members visiting pediatric patients with pertussis and tuberculosis may need to be screened for a history of exposure as well as signs and symptoms of current infection. Potentially infectious visitors are excluded until they receive appropriate medical screening, diagnosis, or treatment. If exclusion is not considered to be in the best interest of the patient or family (i.e., primary family members of critically or terminally ill patients), then the symptomatic visitor must wear a mask while in the healthcare facility and remain in the patient's room, avoiding exposure to others, especially in public waiting areas and the cafeteria.
Visitor screening is used consistently on HSCT units [15, 43]. However, considering the experience during the 2003 SARS outbreaks and the potential for pandemic influenza, developing effective visitor screening systems will be beneficial [9].
Education concerning Respiratory Hygiene/Cough Etiquette is a useful adjunct to visitor screening.

II.N.3.b. Use of barrier precautions by visitors The use of gowns, gloves, or masks by visitors in healthcare settings has not been addressed specifically in the scientific literature. Some studies included the use of gowns and gloves by visitors in the control of MDRO's, but did not perform a separate analysis to determine whether their use by visitors had a measurable impact [893-895]. Family members or visitors who are providing care or having very close patient contact (e.g., feeding, holding) may have contact with other patients and could contribute to transmission if barrier precautions are not used correctly. Specific recommendations may vary by facility or by unit and should be determined by the level of interaction.

Part III:
Precautions to Prevent Transmission of Infectious Agents

There are two tiers of HICPAC/CDC precautions to prevent transmission of infectious agents, Standard Precautions and Transmission-Based Precautions. Standard Precautions are intended to be applied to the care of all patients in all healthcare settings, regardless of the suspected or confirmed presence of an infectious agent. **Implementation of *Standard Precautions* constitutes the primary strategy for the prevention of healthcare-associated transmission of infectious agents among patients and healthcare personnel**.

Transmission-Based Precautions are for patients who are known or suspected to be infected or colonized with infectious agents, including certain epidemiologically important pathogens, which require additional control measures to effectively prevent transmission. Since the infecting agent often is not known at the time of admission to a healthcare facility, Transmission-Based Precautions are used empirically, according to the clinical syndrome and the likely etiologic agents at the time, and then modified when the pathogen is identified or a transmissible infectious etiology is ruled out. Examples of this syndromic approach are presented in Table 2. The HICPAC/CDC Guidelines also include recommendations for creating a Protective Environment for allogeneic HSCT patients.

The specific elements of Standard and Transmission-Based Precautions are discussed in Part II of this guideline. In Part III, the circumstances in which Standard Precautions, Transmission-Based Precautions, and a Protective Environment are applied are discussed. See Tables 4 and 5 for summaries of the key elements of these sets of precautions

III.A. Standard Precautions Standard Precautions combine the major features of Universal Precautions (UP) [780, 896] and Body Substance Isolation (BSI) [640] and are based on the principle that all blood, body fluids, secretions, excretions except sweat, nonintact skin, and mucous membranes may contain transmissible infectious agents. Standard Precautions include a group of infection prevention practices that apply to all patients, regardless of suspected or confirmed infection status, in any setting in which healthcare is delivered (Table 4). These include: hand hygiene; use of gloves, gown, mask, eye protection, or face shield, depending on the anticipated exposure; and safe injection practices. Also, equipment or items in the patient environment likely to have been contaminated with infectious body fluids must be handled in a manner to prevent transmission of infectious agents (e.g. wear gloves for direct contact, contain heavily soiled equipment, properly clean and disinfect or sterilize reusable equipment before use on another patient).

The application of Standard Precautions during patient care is determined by the nature of the HCW-patient interaction and the extent of anticipated blood, body fluid, or pathogen exposure. For some interactions (e.g., performing venipuncture), only gloves may be needed; during other interactions (e.g., intubation), use of gloves, gown, and face shield or mask and goggles is necessary. Education and training on the principles and rationale for

recommended practices are critical elements of Standard Precautions because they facilitate appropriate decision-making and promote adherence when HCWs are faced with new circumstances [655, 681-686]. An example of the importance of the use of Standard Precautions is intubation, especially under emergency circumstances when infectious agents may not be suspected, but later are identified (e.g., SARS-CoV, *N. meningitides*). The application of Standard Precautions is described below and summarized in Table 4. Guidance on donning and removing gloves, gowns and other PPE is presented in the Figure. Standard Precautions are also intended to protect patients by ensuring that healthcare personnel do not carry infectious agents to patients on their hands or via equipment used during patient care.

III.A.1. New Elements of Standard Precautions Infection control problems that are identified in the course of outbreak investigations often indicate the need for new recommendations or reinforcement of existing infection control recommendations to protect patients. Because such recommendations are considered a standard of care and may not be included in other guidelines, they are added here to Standard Precautions. Three such areas of practice that have been added are: Respiratory Hygiene/Cough Etiquette, safe injection practices, and use of masks for insertion of catheters or injection of material into spinal or epidural spaces via lumbar puncture procedures (e.g., myelogram, spinal or epidural anesthesia). While most elements of Standard Precautions evolved from Universal Precautions that were developed for protection of healthcare personnel, these new elements of Standard Precautions focus on protection of patients.

III.A.1.a. Respiratory Hygiene/Cough Etiquette The transmission of SARS-CoV in emergency departments by patients and their family members during the widespread SARS outbreaks in 2003 highlighted the need for vigilance and prompt implementation of infection control measures at the first point of encounter within a healthcare setting (e.g., reception and triage areas in emergency departments, outpatient clinics, and physician offices) [21, 254, 897]. The strategy proposed has been termed Respiratory Hygiene/Cough Etiquette [9, 828] and is intended to be incorporated into infection control practices as a new component of Standard Precautions. The strategy is targeted at patients and accompanying family members and friends with undiagnosed transmissible respiratory infections, and applies to any person with signs of illness including cough, congestion, rhinorrhea, or increased production of respiratory secretions when entering a healthcare facility [40, 41, 43]. The term *cough etiquette* is derived from recommended source control measures for *M. tuberculosis* [12, 126].
The elements of Respiratory Hygiene/Cough Etiquette include 1) education of healthcare facility staff, patients, and visitors; 2) posted signs, in language(s) appropriate to the population served, with instructions to patients and accompanying family members or friends; 3) source control measures (e.g., covering the mouth/nose with a tissue when coughing and prompt disposal of used tissues, using surgical masks on the coughing person when tolerated and

appropriate); 4) hand hygiene after contact with respiratory secretions; and 5) spatial separation, ideally >3 feet, of persons with respiratory infections in common waiting areas when possible. Covering sneezes and coughs and placing masks on coughing patients are proven means of source containment that prevent infected persons from dispersing respiratory secretions into the air [107, 145, 898, 899]. Masking may be difficult in some settings, (e.g., pediatrics, in which case, the emphasis by necessity may be on cough etiquette [900]. Physical proximity of <3 feet has been associated with an increased risk for transmission of infections via the droplet route (e.g., *N. meningitidis* [103] and group A streptococcus [114] and therefore supports the practice of distancing infected persons from others who are not infected. The effectiveness of good hygiene practices, especially hand hygiene, in preventing transmission of viruses and reducing the incidence of respiratory infections both within and outside [901-903] healthcare settings is summarized in several reviews [559, 717, 904].
These measures should be effective in decreasing the risk of transmission of pathogens contained in large respiratory droplets (e.g., influenza virus [23], adenovirus [111], *B. pertussis* [827] and *Mycoplasma pneumoniae* [112]. Although fever will be present in many respiratory infections, patients with pertussis and mild upper respiratory tract infections are often afebrile. Therefore, the absence of fever does not always exclude a respiratory infection. Patients who have asthma, allergic rhinitis, or chronic obstructive lung disease also may be coughing and sneezing. While these patients often are not infectious, cough etiquette measures are prudent.
Healthcare personnel are advised to observe Droplet Precautions (i.e., wear a mask) and hand hygiene when examining and caring for patients with signs and symptoms of a respiratory infection. Healthcare personnel who have a respiratory infection are advised to avoid direct patient contact, especially with high risk patients. If this is not possible, then a mask should be worn while providing patient care.

III.A.1.b. Safe Injection Practices The investigation of four large outbreaks of HBV and HCV among patients in ambulatory care facilities in the United States identified a need to define and reinforce safe injection practices [453]. The four outbreaks occurred in a private medical practice, a pain clinic, an endoscopy clinic, and a hematology/oncology clinic. The primary breaches in infection control practice that contributed to these outbreaks were 1) reinsertion of used needles into a multiple-dose vial or solution container (e.g., saline bag) and 2) use of a single needle/syringe to administer intravenous medication to multiple patients. In one of these outbreaks, preparation of medications in the same workspace where used needle/syringes were dismantled also may have been a contributing factor. These and other outbreaks of viral hepatitis could have been prevented by adherence to basic principles of aseptic technique for the preparation and administration of parenteral medications [453, 454]. These include the use of a sterile, single-use, disposable needle and syringe for each injection given and prevention of contamination of injection equipment and medication.

Whenever possible, use of single-dose vials is preferred over multiple-dose vials, especially when medications will be administered to multiple patients. Outbreaks related to unsafe injection practices indicate that some healthcare personnel are unaware of, do not understand, or do not adhere to basic principles of infection control and aseptic technique. A survey of US healthcare workers who provide medication through injection found that 1% to 3% reused the same needle and/or syringe on multiple patients [905]. Among the deficiencies identified in recent outbreaks were a lack of oversight of personnel and failure to follow-up on reported breaches in infection control practices in ambulatory settings. Therefore, to ensure that all healthcare workers understand and adhere to recommended practices, principles of infection control and aseptic technique need to be reinforced in training programs and incorporated into institutional polices that are monitored for adherence [454].

III.A.1.c. Infection Control Practices for Special Lumbar Puncture Procedues In 2004, CDC investigated eight cases of post-myelography meningitis that either were reported to CDC or identified through a survey of the Emerging Infections Network of the Infectious Disease Society of America. Blood and/or cerebrospinal fluid of all eight cases yielded streptococcal species consistent with oropharyngeal flora and there were changes in the CSF indices and clinical status indicative of bacterial meningitis. Equipment and products used during these procedures (e.g., contrast media) were excluded as probable sources of contamination. Procedural details available for seven cases determined that antiseptic skin preparations and sterile gloves had been used. However, none of the clinicians wore a face mask, giving rise to the speculation that droplet transmission of oralpharyngeal flora was the most likely explanation for these infections. Bacterial meningitis following myelogram and other spinal procedures (e.g., lumbar puncture, spinal and epidural anesthesia, intrathecal chemotherapy) has been reported previously [906-915]. As a result, the question of whether face masks should be worn to prevent droplet spread of oral flora during spinal procedures (e.g., myelogram, lumbar puncture, spinal anesthesia) has been debated [916, 917]. Face masks are effective in limiting the dispersal of oropharyngeal droplets [918] and are recommended for the placement of central venous catheters [919]. In October 2005, the Healthcare Infection Control Practices Advisory Committee (HICPAC) reviewed the evidence and concluded that there is sufficient experience to warrant the additional protection of a face mask for the individual placing a catheter or injecting material into the spinal or epidural space.

III.B. Transmission-Based Precautions There are three categories of Transmission-Based Precautions: Contact Precautions, Droplet Precautions, and Airborne Precautions. Transmission-Based Precautions are used when the route(s) of transmission is (are) not completely interrupted using Standard Precautions alone. For some diseases that have multiple routes of transmission (e.g., SARS), more than one Transmission-Based Precautions category may be used. When used either singly or in combination, they are always used in

addition to Standard Precautions. See Appendix A for recommended precautions for specific infections. When Transmission-Based Precautions are indicated, efforts must be made to counteract possible adverse effects on patients (i.e., anxiety, depression and other mood disturbances [920-922], perceptions of stigma [923], reduced contact with clinical staff [924-926], and increases in preventable adverse events [565] in order to improve acceptance by the patients and adherence by HCWs.

III.B.1. Contact Precautions Contact Precautions are intended to prevent transmission of infectious agents, including epidemiologically important microorganisms, which are spread by direct or indirect contact with the patient or the patient's environment as described in I.B.3.a. The specific agents and circumstance for which Contact Precautions are indicated are found in Appendix A. The application of Contact Precautions for patients infected or colonized with MDROs is described in the 2006 HICPAC/CDC MDRO guideline [927]. Contact Precautions also apply where the presence of excessive wound drainage, fecal incontinence, or other discharges from the body suggest an increased potential for extensive environmental contamination and risk of transmission. A single-patient room is preferred for patients who require Contact Precautions. When a single-patient room is not available, consultation with infection control personnel is recommended to assess the various risks associated with other patient placement options (e.g., cohorting, keeping the patient with an existing roommate). In multi-patient rooms, $\geq$3 feet spatial separation between beds is advised to reduce the opportunities for inadvertent sharing of items between the infected/colonized patient and other patients. Healthcare personnel caring for patients on Contact Precautions wear a gown and gloves for all interactions that may involve contact with the patient or potentially contaminated areas in the patient's environment. Donning PPE upon room entry and discarding before exiting the patient room is done to contain pathogens, especially those that have been implicated in transmission through environmental contamination (e.g., VRE, *C. difficile,* noroviruses and other intestinal tract pathogens; RSV) [54, 72, 73, 78, 274, 275, 740].

III.B.2. Droplet Precautions Droplet Precautions are intended to prevent transmission of pathogens spread through close respiratory or mucous membrane contact with respiratory secretions as described in I.B.3.b. Because these pathogens do not remain infectious over long distances in a healthcare facility, special air handling and ventilation are not required to prevent droplet transmission. Infectious agents for which Droplet Precautions are indicated are found in Appendix A and include *B. pertussis*, influenza virus, adenovirus, rhinovirus, *N. meningitides*, and group A streptococcus (for the first 24 hours of antimicrobial therapy). A single patient room is preferred for patients who require Droplet Precautions. When a single-patient room is not available, consultation with infection control personnel is recommended to assess the various risks associated with other patient placement options (e.g., cohorting, keeping the patient with an existing roommate). Spatial separation of $\geq$ 3 feet and drawing

the curtain between patient beds is especially important for patients in multi-bed rooms with infections transmitted by the droplet route. Healthcare personnel wear a mask (a respirator is not necessary) for close contact with infectious patient; the mask is generally donned upon room entry. Patients on Droplet Precautions who must be transported outside of the room should wear a mask if tolerated and follow Respiratory Hygiene/Cough Etiquette.

III.B.3. Airborne Precautions Airborne Precautions prevent transmission of infectious agents that remain infectious over long distances when suspended in the air (e.g., rubeola virus [measles], varicella virus [chickenpox], *M. tuberculosis,* and possibly SARS-CoV) as described in I.B.3.c and Appendix A. The preferred placement for patients who require Airborne Precautions is in an airborne infection isolation room (AIIR). An AIIR is a single-patient room that is equipped with special air handling and ventilation capacity that meet the American Institute of Architects/Facility Guidelines Institute (AIA/FGI) standards for AIIRs (i.e., monitored negative pressure relative to the surrounding area, 12 air exchanges per hour for new construction and renovation and 6 air exchanges per hour for existing facilities, air exhausted directly to the outside or recirculated through HEPA filtration before return) [12, 13]. Some states require the availability of such rooms in hospitals, emergency departments, and nursing homes that care for patients with *M. tuberculosis*. A respiratory protection program that includes education about use of respirators, fit-testing, and user seal checks is required in any facility with AIIRs. In settings where Airborne Precautions cannot be implemented due to limited engineering resources (e.g., physician offices), masking the patient, placing the patient in a private room (e.g., office examination room) with the door closed, and providing N95 or higher level respirators or masks if respirators are not available for healthcare personnel will reduce the likelihood of airborne transmission until the patient is either transferred to a facility with an AIIR or returned to the home environment, as deemed medically appropriate. Healthcare personnel caring for patients on Airborne Precautions wear a mask or respirator, depending on the disease-specific recommendations (Respiratory Protection II.E.4, Table 2, and Appendix A), that is donned prior to room entry. Whenever possible, non-immune HCWs should not care for patients with vaccine-preventable airborne diseases (e.g., measles, chickenpox, and smallpox).

III.C. Syndromic and empiric applications of Transmission-Based Precautions Diagnosis of many infections requires laboratory confirmation. Since laboratory tests, especially those that depend on culture techniques, often require two or more days for completion, Transmission-Based Precautions must be implemented while test results are pending based on the clinical presentation and likely pathogens. Use of appropriate Transmission-Based Precautions at the time a patient develops symptoms or signs of transmissible infection, or arrives at a healthcare facility for care, reduces transmission opportunities. While it is not possible to identify prospectively all patients needing Transmission-Based Precautions, certain clinical syndromes and conditions carry a sufficiently high

risk to warrant their use empirically while confirmatory tests are pending (Table 2). Infection control professionals are encouraged to modify or adapt this table according to local conditions.

III.D. Discontinuation of Transmission-Based Precautions Transmission-Based Precautions remain in effect for limited periods of time (i.e., while the risk for transmission of the infectious agent persists or for the duration of the illness (Appendix A). For most infectious diseases, this duration reflects known patterns of persistence and shedding of infectious agents associated with the natural history of the infectious process and its treatment. For some diseases (e.g., pharyngeal or cutaneous diphtheria, RSV), Transmission-Based Precautions remain in effect until culture or antigen-detection test results document eradication of the pathogen and, for RSV, symptomatic disease is resolved. For other diseases, (e.g., *M. tuberculosis*) state laws and regulations, and healthcare facility policies, may dictate the duration of precautions[12]). In immunocompromised patients, viral shedding can persist for prolonged periods of time (many weeks to months) and transmission to others may occur during that time; therefore, the duration of contact and/or droplet precautions may be prolonged for many weeks [500, 928-933].

The duration of Contact Precautions for patients who are colonized or infected with MDROs remains undefined. MRSA is the only MDRO for which effective decolonization regimens are available [867]. However, carriers of MRSA who have negative nasal cultures after a course of systemic or topical therapy may resume shedding MRSA in the weeks that follow therapy [934, 935]. Although early guidelines for VRE suggested discontinuation of Contact Precautions after three stool cultures obtained at weekly intervals proved negative [740], subsequent experiences have indicated that such screening may fail to detect colonization that can persist for >1 year [27, 936-938]. Likewise, available data indicate that colonization with VRE, MRSA [939], and possibly MDR-GNB, can persist for many months, especially in the presence of severe underlying disease, invasive devices, and recurrent courses of antimicrobial agents.

It may be prudent to assume that MDRO carriers are colonized permanently and manage them accordingly. Alternatively, an interval free of hospitalizations, antimicrobial therapy, and invasive devices (e.g., 6 or 12 months) before reculturing patients to document clearance of carriage may be used. Determination of the best strategy awaits the results of additional studies. See the 2006 HICPAC/CDC MDRO guideline [927] for discussion of possible criteria to discontinue Contact Precautions for patients colonized or infected with MDROs.

III.E. Application of Transmission-Based Precautions in ambulatory and home care settings Although Transmission-Based Precautions generally apply in all healthcare settings, exceptions exist. For example, in home care, AIIRs are not available. Furthermore, family members already exposed to diseases such as varicella and tuberculosis would not use masks or respiratory protection, but visiting HCWs would need to use such protection. Similarly, management of patients colonized or infected with MDROs may necessitate

Contact Precautions in acute care hospitals and in some LTCFs when there is continued transmission, but the risk of transmission in ambulatory care and home care, has not been defined. Consistent use of Standard Precautions may suffice in these settings, but more information is needed.

III.F. Protective Environment A Protective Environment is designed for allogeneic HSCT patients to minimize fungal spore counts in the air and reduce the risk of invasive environmental fungal infections (see Table 5 for specifications) [11, 13-15]. The need for such controls has been demonstrated in studies of aspergillus outbreaks associated with construction [11, 14, 15, 157, 158]. As defined by the American Insitute of Architecture [13] and presented in detail in the Guideline for Environmental Infection Control 2003 [11, 861], air quality for HSCT patients is improved through a combination of environmental controls that include 1) HEPA filtration of incoming air; 2) directed room air flow; 3) positive room air pressure relative to the corridor; 4) well-sealed rooms (including sealed walls, floors, ceilings, windows, electrical outlets) to prevent flow of air from the outside; 5) ventilation to provide $\geq$12 air changes per hour; 6) strategies to minimize dust (e.g., scrubbable surfaces rather than upholstery [940] and carpet [941], and routinely cleaning crevices and sprinkler heads); and 7) prohibiting dried and fresh flowers and potted plants in the rooms of HSCT patients. The latter is based on molecular typing studies that have found indistinguishable strains of *Aspergillus terreus* in patients with hematologic malignancies and in potted plants in the vicinity of the patients [942-944]. The desired quality of air may be achieved without incurring the inconvenience or expense of laminar airflow [15, 157]. To prevent inhalation of fungal spores during periods when construction, renovation, or other dust-generating activities that may be ongoing in and around the health-care facility, it has been advised that severely immunocompromised patients wear a high-efficiency respiratory-protection device (e.g., an N95 respirator) when they leave the Protective Environment [11, 14, 945]). The use of masks or respirators by HSCT patients when they are outside of the Protective Environment for prevention of environmental fungal infections in the absence of construction has not been evaluated. A Protective Environment does not include the use of barrier precautions beyond those indicated for Standard and Transmission-Based Precautions. No published reports support the benefit of placing solid organ transplants or other immunocompromised patients in a Protective Environment.

Part IV:

Recommendations

These recommendations are designed to prevent transmission of infectious agents among patients and healthcare personnel in all settings where healthcare is delivered. As in other CDC/HICPAC guidelines, each recommendation is categorized on the basis of existing scientific data, theoretical rationale, applicability, and when possible, economic impact. The CDC/HICPAC system for categorizing recommendations is as follows:

Category IA Strongly recommended for implementation and strongly supported by well-designed experimental, clinical, or epidemiologic studies.

Category IB Strongly recommended for implementation and supported by some experimental, clinical, or epidemiologic studies and a strong theoretical rationale.

Category IC Required for implementation, as mandated by federal and/or state regulation or standard.

Category II Suggested for implementation and supported by suggestive clinical or epidemiologic studies or a theoretical rationale.

No recommendation; unresolved issue. Practices for which insufficient evidence or no consensus regarding efficacy exists.

I. Administrative Responsibilities

Healthcare organization administrators should ensure the implementation of recommendations in this section.

I.A. Incorporate preventing transmission of infectious agents into the objectives of the organization's patient and occupational safety programs [543-546, 561, 620, 626, 946]. *Category IB/IC*

I.B. Make preventing transmission of infectious agents a priority for the healthcare organization. Provide administrative support, including fiscal and human resources for maintaining infection control programs [434, 548, 549, 559, 561, 566, 662 552, 562-564, 946]. *Category IB/IC*

I.B.1. Assure that individuals with training in infection control are employed by or are available by contract to all healthcare facilities so that the infection control program is managed by one or more qualified individuals [552, 566 316, 575, 947 573, 576, 946]. *Category IB/IC*

I.B.1.a. Determine the specific infection control full-time equivalents (FTEs) according to the scope of the infection control program, the complexity of the healthcare facility or system, the characteristics of the patient population, the unique or urgent needs of the facility and community, and proposed staffing levels based on survey results and recommendations from professional organizations [434, 549 552, 566 316, 569, 573, 575 948 949]. *Category IB*

I.B.2. Include prevention of healthcare-associated infections (HAI) as one determinant of bedside nurse staffing levels and composition,

especially in high-risk units [585-589] [590] [592] [593] [551, 594, 595] [418, 596, 597] [583]. *Category IB*

I.B.3. Delegate authority to infection control personnel or their designees (e.g., patient care unit charge nurses) for making infection control decisions concerning patient placement and assignment of Transmission-Based Precautions [549] [434, 857, 946]. *Category IC*

I.B.4. Involve infection control personnel in decisions on facility construction and design, determination of AIIR and Protective Environment capacity needs and environmental assessments [11, 13, 950] [951] [12]. *Category IB/IC*

I.B.4.a. Provide ventilation systems required for a sufficient number of AIIRs (as determined by a risk assessment) and Protective Environments in healthcare facilities that provide care to patients for whom such rooms are indicated, according to published recommendations [11-13, 15]. *Category IB/IC*

I.B.5. Involve infection control personnel in the selection and post-implementation evaluation of medical equipment and supplies and changes in practice that could affect the risk of HAI [952, 953]. *Category IC*

I.B.6. Ensure availability of human and fiscal resources to provide clinical microbiology laboratory support, including a sufficient number of medical technologists trained in microbiology, appropriate to the healthcare setting, for monitoring transmission of microorganisms, planning and conducting epidemiologic investigations, and detecting emerging pathogens. Identify resources for performing surveillance cultures, rapid diagnostic testing for viral and other selected pathogens, preparation of antimicrobial susceptibility summary reports, trend analysis, and molecular typing of clustered isolates (performed either on-site or in a reference laboratory) and use these resources according to facility-specific epidemiologic needs, in consultation with clinical microbiologists [553, 609, 610, 612, 617, 954] [614] [603, 615, 616] [605] [599] [554] [598, 606, 607]. *Category IB*

I.B.7. Provide human and fiscal resources to meet occupational health needs related to infection control (e.g., healthcare personnel immunization, post-exposure evaluation and care, evaluation and management of healthcare personnel with communicable infections [739] [12] [17, 879-881, 955] [134] [690]. *Category IB/IC*

I.B.8. In all areas where healthcare is delivered, provide supplies and equipment necessary for the consistent observance of Standard Precautions, including hand hygiene products and personal protective equipment (e.g., gloves, gowns, face and eye protection) [739] [559] [946]. *Category IB/IC*

I.B.9. Develop and implement policies and procedures to ensure that reusable patient care equipment is cleaned and reprocessed appropriately before use on another patient [11, 956] [957, 958] [959] [836] [87] [11, 960] [961]. *Category IA/IC*

I.C. Develop and implement processes to ensure oversight of infection control activities appropriate to the healthcare setting and assign responsibility for oversight of infection control activities to an individual or group within the healthcare organization that is knowledgeable about infection control [434, 549, 566]. *Category II*

I.D. Develop and implement systems for early detection and management (e.g., use of appropriate infection control measures, including isolation precautions, PPE) of potentially infectious persons at initial points of patient encounter in outpatient settings (e.g., triage areas, emergency departments, outpatient clinics, physician offices) and at the time of admission to hospitals and long-term care facilities (LTCF) [9, 122, 134, 253, 827]. *Category IB*

I.E. Develop and implement policies and procedures to limit patient visitation by persons with signs or symptoms of a communicable infection. Screen visitors to high-risk patient care areas (e.g., oncology units, hematopoietic stem call transplant [HSCT] units, intensive care units, other severely immunocompromised patients) for possible infection [43 24, 41, 962, 963].*Category IB*

I.F. Identify performance indicators of the effectiveness of organization-specific measures to prevent transmission of infectious agents (Standard and Transmission-Based Precautions), establish processes to monitor adherence to those performance measures and provide feedback to staff members [704 739 705 708 666, 964 667 668 555]. *Category IB*

II. Education and Training

II.A. Provide job- or task-specific education and training on preventing transmission of infectious agents associated with healthcare during orientation to the healthcare facility; update information periodically during ongoing education programs. Target all healthcare personnel for education and training, including but not limited to medical, nursing, clinical technicians, laboratory staff; property service (housekeeping), laundry, maintenance and dietary workers; students, contract staff and volunteers. Document competency initially and repeatedly, as appropriate, for the specific staff positions. Develop a system to ensure that healthcare personnel employed by outside agencies meet these education and training requirements through programs offered by the agencies or by participation in the healthcare facility's program designed for full-time personnel [126, 559, 561, 562, 655, 681-684, 686, 688, 689, 702, 893, 919, 965]. *Category IB*

II.A.1. Include in education and training programs, information concerning use of vaccines as an adjunctive infection control measure [17, 611, 690, 874]. *Category IB*

II.A.2. Enhance education and training by applying principles of adult learning, using reading level and language appropriate material for the target audience, and using online educational tools available to the institution [658, 694, 695, 697, 698, 700, 966]. *Category IB*

II.B. Provide instructional materials for patients and visitors on recommended hand hygiene and Respiratory Hygiene/Cough Etiquette practices and the application of Transmission-Based Precautions [9, 709, 710, 963]. *Category II*

III. Surveillance

III.A. Monitor the incidence of epidemiologically-important organisms and targeted HAIs that have substantial impact on outcome and for which effective preventive interventions are available; use information collected through surveillance of high-risk populations, procedures, devices and highly transmissible infectious agents to detect transmission of infectious agents in the healthcare facility [566, 671, 672, 675, 687, 919, 967, 968 673 969 970]. *Category IA*

III.B. Apply the following epidemiologic principles of infection surveillance [671, 967 673 969 663 664]. *Category IB*

- Use standardized definitions of infection
- Use laboratory-based data (when available)
- Collect epidemiologically-important variables (e.g., patient locations and/or clinical service in hospitals and other large multi-unit facilities, population-specific risk factors [e.g., low birth-weight neonates], underlying conditions that predispose to serious adverse outcomes)
- Analyze data to identify trends that may indicated increased rates of transmission
- Feedback information on trends in the incidence and prevalence of HAIs, probable risk factors, and prevention strategies and their impact to the appropriate healthcare providers, organization administrators, and as required by local and state health authorities

III.C. Develop and implement strategies to reduce risks for transmission and evaluate effectiveness [566, 673, 684, 970 963 971]. *Category IB*

III.D. When transmission of epidemiologically-important organisms continues despite implementation and documented adherence to infection prevention and control strategies, obtain consultation from persons knowledgeable in infection control and healthcare epidemiology to review the situation and recommend additional measures for control [566 247 687]. *Category IB*

III.E. Review periodically information on community or regional trends in the incidence and prevalence of epidemiologically-important organisms (e.g., influenza, RSV, pertussis, invasive group A streptococcal disease, MRSA, VRE) (including in other healthcare facilities) that may impact transmission of organisms within the facility [398, 687, 972, 973 974]. *Category II*

IV. Standard Precautions

Assume that every person is potentially infected or colonized with an organism that could be transmitted in the healthcare setting and apply the following infection control practices during the delivery of health care.

IV.A. Hand Hygiene

IV.A.1. During the delivery of healthcare, avoid unnecessary touching of surfaces in close proximity to the patient to prevent both contamination of clean hands from environmental surfaces and transmission of pathogens from contaminated hands to surfaces[72, 73 739, 800, 975{CDC, 2001 #970]. *Category IB/IC*

IV.A.2. When hands are visibly dirty, contaminated with proteinaceous material, or visibly soiled with blood or body fluids, wash hands with either a nonantimicrobial soap and water or an antimicrobial soap and water [559]. *Category IA*

IV.A.3. If hands are not visibly soiled, or after removing visible material with nonantimicrobial soap and water, decontaminate hands in the clinical situations described in IV.A.2.a-f. The preferred method of hand decontamination is with an alcohol-based hand rub [562, 978]. Alternatively, hands may be washed with an antimicrobial soap and water. Frequent use of alcohol-based hand rub immediately following handwashing with nonantimicrobial soap may increase the frequency of dermatitis [559]. *Category IB*

Perform hand hygiene:

IV.A.3.a. Before having direct contact with patients [664, 979]. *Category IB*

IV.A.3.b. After contact with blood, body fluids or excretions, mucous membranes, nonintact skin, or wound dressings [664]. *Category IA*

IV.A.3.c. After contact with a patient's intact skin (e.g., when taking a pulse or blood pressure or lifting a patient) [167, 976, 979, 980]. *Category IB*

IV.A.3.d. If hands will be moving from a contaminated-body site to a clean-body site during patient care. *Category II*

IV.A.3.e. After contact with inanimate objects (including medical equipment) in the immediate vicinity of the patient [72, 73, 88, 800, 981 982]. *Category II*

IV.A.3.f. After removing gloves [728, 741, 742]. *Category IB*

IV.A.4. Wash hands with non-antimicrobial soap and water or with antimicrobial soap and water if contact with spores (e.g., *C. difficile* or *Bacillus anthracis*) is likely to have occurred. The physical action of washing and rinsing hands under such circumstances is recommended because alcohols, chlorhexidine, iodophors, and other antiseptic agents have poor activity against spores [559, 956, 983]. *Category II*

IV.A.5. Do not wear artificial fingernails or extenders if duties include direct contact with patients at high risk for infection and associated adverse outcomes (e.g., those in ICUs or operating rooms) [30, 31, 559, 722-724]. *Category IA*

IV.A.5.a. Develop an organizational policy on the wearing of non-natural nails by healthcare personnel who have direct contact with patients outside of the groups specified above [984]. *Category II*

IV.B. Personal protective equipment (PPE) (see Figure)

IV.B.1. Observe the following principles of use:

IV.B.1.a. Wear PPE, as described in IV.B.2-4,when the nature of the anticipated patient interaction indicates that contact with blood or body fluids may occur [739, 780, 896]. *Category IB/IC*

IV.B.1.b. Prevent contamination of clothing and skin during the process of removing PPE (see Figure). *Category II*

IV.B.1.c. Before leaving the patient's room or cubicle, remove and discard PPE [18, 739]. *Category IB/IC*

IV.B.2. Gloves

IV.B.2.a. Wear gloves when it can be reasonably anticipated that contact with blood or other potentially infectious materials, mucous membranes, nonintact skin, or potentially contaminated intact skin (e.g., of a patient incontinent of stool or urine) could occur [18, 728, 739, 741, 780, 985]. *Category IB/IC*

IV.B.2.b. Wear gloves with fit and durability appropriate to the task [559, 731, 732, 739, 986, 987]. *Category IB*

IV.B.2.b.i. Wear disposable medical examination gloves for providing direct patient care.

IV.B.2.b.ii. Wear disposable medical examination gloves or reusable utility gloves for cleaning the environment or medical equipment.

IV.B.2.c. Remove gloves after contact with a patient and/or the surrounding environment (including medical equipment) using proper technique to prevent hand contamination (see Figure). Do not wear the same pair of gloves for the care of more than one patient. Do not wash gloves for the purpose of reuse since this practice has been associated with transmission of pathogens [559, 728, 741-743, 988]. *Category IB*

IV.B.2.d. Change gloves during patient care if the hands will move from a contaminated body-site (e.g., perineal area) to a clean body-site (e.g., face). *Category II*

IV.B.3. Gowns

IV.B.3.a. Wear a gown, that is appropriate to the task, to protect skin and prevent soiling or contamination of clothing during procedures and patient-care activities when contact with blood, body fluids, secretions, or excretions is anticipated [739, 780, 896]. *Category IB/IC*

IV.B.3.a.i. Wear a gown for direct patient contact if the patient has uncontained secretions or excretions [24, 88, 89, 739, 744] *Category IB/IC*

IV.B.3.a.ii. Remove gown and perform hand hygiene before leaving the patient's environment [24, 88, 89, 739, 744] *Category IB/IC*

IV.B.3.b. Do not reuse gowns, even for repeated contacts with the same patient. *Category II*

IV.B.3.c. Routine donning of gowns upon entrance into a high risk unit (e.g., ICU, NICU, HSCT unit) is not indicated [365, 747-750]. *Category IB*

IV.B.4. Mouth, nose, eye protection

IV.B.4.a. Use PPE to protect the mucous membranes of the eyes, nose and mouth during procedures and patient-care activities that are likely to generate splashes or sprays of blood, body fluids, secretions and excretions. Select masks, goggles, face shields, and combinations of each according to the need anticipated by the task performed [113, 739, 780, 896]. *Category IB/IC*

IV.B.5. During aerosol-generating procedures (e.g., bronchoscopy, suctioning of the respiratory tract [if not using in-line suction catheters], endotracheal intubation) in patients who are not suspected of being infected with an agent for which respiratory protection is otherwise recommended (e.g., *M. tuberculosis,* SARS or hemorrhagic fever viruses), wear one of the following: a face shield that fully covers the front and sides of the face, a mask with attached shield, or a mask and goggles (in addition to gloves and gown) [95, 96, 113, 126 93 94, 134]. *Category IB*

IV.C. Respiratory Hygiene/Cough Etiquette

IV.C.1. Educate healthcare personnel on the importance of source control measures to contain respiratory secretions to prevent droplet and fomite transmission of respiratory pathogens, especially during seasonal outbreaks of viral respiratory tract infections (e.g., influenza, RSV, adenovirus, parainfluenza virus) in communities [14, 24, 684 10, 262]. *Category IB*

IV.C.2. Implement the following measures to contain respiratory secretions in patients and accompanying individuals who have signs and symptoms of a respiratory infection, beginning at the point of initial encounter in a healthcare setting (e.g., triage, reception and waiting areas in emergency departments, outpatient clinics and physician offices) [20, 24, 145, 902, 989].

IV.C.2.a. Post signs at entrances and in strategic places (e.g., elevators, cafeterias) within ambulatory and inpatient settings with instructions to patients and other persons with symptoms of a respiratory infection to cover their mouths/noses when coughing or sneezing, use and dispose of tissues, and perform hand hygiene after hands have been in contact with respiratory secretions. *Category II*

IV.C.2.b. Provide tissues and no-touch receptacles (e.g.,foot-pedal-operated lid or open, plastic-lined waste basket) for disposal of tissues [20]. *Category II*

IV.C.2.c. Provide resources and instructions for performing hand hygiene in or near waiting areas in ambulatory and inpatient settings; provide conveniently-located dispensers of alcohol-based hand rubs and, where sinks are available, supplies for handwashing [559, 903]. *Category IB*

IV.C.2.d. During periods of increased prevalence of respiratory infections in the community (e.g., as indicated by increased school absenteeism, increased number of patients seeking care for a

respiratory infection), offer masks to coughing patients and other symptomatic persons (e.g., persons who accompany ill patients) upon entry into the facility or medical office [126, 899 898] and encourage them to maintain special separation, ideally a distance of at least 3 feet, from others in common waiting areas [23, 103, 111, 114 20, 134]. *Category IB*

IV.C.2.d.i. Some facilities may find it logistically easier to institute this recommendation year-round as a standard of practice. *Category II*

IV.D. Patient placement

IV.D.1. Include the potential for transmission of infectious agents in patient-placement decisions. Place patients who pose a risk for transmission to others (e.g., uncontained secretions, excretions or wound drainage; infants with suspected viral respiratory or gastrointestinal infections) in a single-patient room when available [24, 430, 435, 796, 797, 806, 990 410, 793]. *Category IB*

IV.D.2. Determine patient placement based on the following principles:

- Route(s) of transmission of the known or suspected infectious agent
- Risk factors for transmission in the infected patient
- Risk factors for adverse outcomes resulting from an HAI in other patients in the area or room being considered for patient-placement
- Availability of single-patient rooms
- Patient options for room-sharing (e.g., cohorting patients with the same infection) *Category II*

IV.E. Patient-care equipment and instruments/devices [956]

IV.E.1. Establish policies and procedures for containing, transporting, and handling patient-care equipment and instruments/devices that may be contaminated with blood or body fluids [18, 739, 975]. *Category IB/IC*

IV.E.2. Remove organic material from critical and semi-critical instrument/devices, using recommended cleaning agents before high level disinfection and sterilization to enable effective disinfection and sterilization processes [836 991, 992]. *Category IA*

IV.E.3. Wear PPE (e.g., gloves, gown), according to the level of anticipated contamination, when handling patient-care equipment and instruments/devices that is visibly soiled or may have been in contact with blood or body fluids [18, 739, 975]. *Category IB/IC*

IV.F. Care of the environment [11]

IV.F.1. Establish policies and procedures for routine and targeted cleaning of environmental surfaces as indicated by the level of patient contact and degree of soiling [11]. *Category II*

IV.F.2. Clean and disinfect surfaces that are likely to be contaminated with pathogens, including those that are in close proximity to the patient (e.g., bed rails, over bed tables) and frequently-touched surfaces in the patient care environment (e.g., door knobs, surfaces in and

surrounding toilets in patients' rooms) on a more frequent schedule compared to that for other surfaces (e.g., horizontal surfaces in waiting rooms) [11 73, 740, 746, 993, 994 72, 800, 835 995]. *Category IB*

IV.F.3. Use EPA-registered disinfectants that have microbiocidal (i.e., killing) activity against the pathogens most likely to contaminate the patient-care environment. Use in accordance with manufacturer's instructions [842-844, 956, 996]. *Category IB/IC*

IV.F.3.a. Review the efficacy of in-use disinfectants when evidence of continuing transmission of an infectious agent (e.g., rotavirus, *C. difficile,* norovirus) may indicate resistance to the in-use product and change to a more effective disinfectant as indicated [275, 842, 847]. *Category II*

IV.F.4. In facilities that provide health care to pediatric patients or have waiting areas with child play toys (e.g., obstetric/gynecology offices and clinics), establish policies and procedures for cleaning and disinfecting toys at regular intervals [379 80]. *Category IB*

- Use the following principles in developing this policy and procedures: *Category II*
  - Select play toys that can be easily cleaned and disinfected
  - Do not permit use of stuffed furry toys if they will be shared
  - Clean and disinfect large stationary toys (e.g., climbing equipment) at least weekly and whenever visibly soiled
  - If toys are likely to be mouthed, rinse with water after disinfection; alternatively wash in a dishwasher
  - When a toy requires cleaning and disinfection, do so immediately or store in a designated labeled container separate from toys that are clean and ready for use

IV.F.5. Include multi-use electronic equipment in policies and procedures for preventing contamination and for cleaning and disinfection, especially those items that are used by patients, those used during delivery of patient care, and mobile devices that are moved in and out of patient rooms frequently (e.g., daily) [850 851, 852, 997]. *Category IB*

IV.F.5.a. No recommendation for use of removable protective covers or washable keyboards. *Unresolved issue*

IV.G. Textiles and laundry

IV.G.1. Handle used textiles and fabrics with minimum agitation to avoid contamination of air, surfaces and persons [739, 998, 999]. *Category IB/IC*

IV.G.2. If laundry chutes are used, ensure that they are properly designed, maintained, and used in a manner to minimize dispersion of aerosols from contaminated laundry [11, 13, 1000, 1001]. *Category IB/IC*

IV.H. Safe injection practices

The following recommendations apply to the use of needles, cannulas that replace needles, and, where applicable intravenous delivery systems [454]

IV.H.1. Use aseptic technique to avoid contamination of sterile injection equipment [1002, 1003]. *Category IA*

IV.H.2. Do not administer medications from a syringe to multiple patients, even if the needle or cannula on the syringe is changed. Needles, cannulae and syringes are sterile, single-use items; they should not be reused for another patient nor to access a medication or solution that might be used for a subsequent patient [453, 919, 1004, 1005]. *Category IA*

IV.H.3. Use fluid infusion and administration sets (i.e., intravenous bags, tubing and connectors) for one patient only and dispose appropriately after use. Consider a syringe or needle/cannula contaminated once it has been used to enter or connect to a patient's intravenous infusion bag or administration set [453]. *Category IB*

IV.H.4. Use single-dose vials for parenteral medications whenever possible [453]. *Category IA*

IV.H.5. Do not administer medications from single-dose vials or ampules to multiple patients or combine leftover contents for later use [369 453, 1005]. *Category IA*

IV.H.6. If multidose vials must be used, both the needle or cannula and syringe used to access the multidose vial must be sterile [453, 1002]. *Category IA*

IV.H.7. Do not keep multidose vials in the immediate patient treatment area and store in accordance with the manufacturer's recommendations; discard if sterility is compromised or questionable [453, 1003]. *Category IA*

IV.H.8. Do not use bags or bottles of intravenous solution as a common source of supply for multiple patients [453, 1006]. *Category IB*

IV.I. Infection control practices for special lumbar puncture procedures

Wear a surgical mask when placing a catheter or injecting material into the spinal canal or subdural space (i.e., during myelograms, lumbar puncture and spinal or epidural anesthesia [906 907-909 910, 911 912-914, 918 1007]. *Category IB*

IV.J. Worker safety

Adhere to federal and state requirements for protection of healthcare personnel from exposure to bloodborne pathogens [739]. *Category IC*

V. Transmission-Based Precautions

V.A. General principles

V.A.1. In addition to Standard Precautions, use Transmission-Based Precautions for patients with documented or suspected infection or colonization with highly transmissible or epidemiologically-important pathogens for which additional precautions are needed to prevent transmission (see Appendix A) [24, 93, 126, 141, 306, 806, 1008]. *Category IA*

V.A.2. Extend duration of Transmission-Based Precautions, (e.g., Droplet, Contact) for immunosuppressed patients with viral infections due to

prolonged shedding of viral agents that may be transmitted to others [928, 931-933, 1009-1011]. *Category IA*

V.B. Contact Precautions

V.B.1. Use Contact Precautions as recommended in Appendix A for patients with known or suspected infections or evidence of syndromes that represent an increased risk for contact transmission. For specific recommendations for use of Contact Precautions for colonization or infection with MDROs, go to the MDRO guideline: www.cdc.gov/ncidod/dhqp/pdf/ar/mdroGuideline2006.pdf [870].

V.B.2. Patient placement

V.B.2.a. In *acute care hospitals*, place patients who require Contact Precautions in a single-patient room when available [24, 687, 793, 796, 797, 806, 837, 893, 1012, 1013] *Category IB*

When single-patient rooms are in short supply, apply the following principles for making decisions on patient placement:

- Prioritize patients with conditions that may facilitate transmission (e.g., uncontained drainage, stool incontinence) for single-patient room placement. *Category II*
- Place together in the same room (cohort) patients who are infected or colonized with the same pathogen and are suitable roommates [29, 638, 808, 811-813, 815, 818, 819] *Category IB*
- If it becomes necessary to place a patient who requires Contact Precautions in a room with a patient who is not infected or colonized with the same infectious agent:
    - Avoid placing patients on Contact Precautions in the same room with patients who have conditions that may increase the risk of adverse outcome from infection or that may facilitate transmission (e.g., those who are immunocompromised, have open wounds, or have anticipated prolonged lengths of stay). *Category II*
    - Ensure that patients are physically separated (i.e., >3 feet apart) from each other. Draw the privacy curtain between beds to minimize opportunities for direct contact.) *Category II*
    - Change protective attire and perform hand hygiene between contact with patients in the same room, regardless of whether one or both patients are on Contact Precautions [728, 741, 742, 988, 1014, 1015]. *Category IB*

V.B.2.b. In *long-term care and other residential settings*, make decisions regarding patient placement on a case-by-case basis, balancing infection risks to other patients in the room, the presence of risk factors that increase the likelihood of transmission, and the

potential adverse psychological impact on the infected or colonized patient [920, 921]. *Category II*

V.B.2.c. In *ambulatory settings*, place patients who require Contact Precautions in an examination room or cubicle as soon as possible [20]. *Category II*

V.B.3. Use of personal protective equipment

V.B.3.a. Gloves

Wear gloves whenever touching the patient's intact skin [24, 89, 134, 559, 746, 837] or surfaces and articles in close proximity to the patient (e.g., medical equipment, bed rails) [72, 73, 88, 837]. Don gloves upon entry into the room or cubicle. *Category IB*

V.B.3.b. Gowns

V.B.3.b.i. Wear a gown whenever anticipating that clothing will have direct contact with the patient or potentially contaminated environmental surfaces or equipment in close proximity to the patient. Don gown upon entry into the room or cubicle. Remove gown and observe hand hygiene before leaving the patient-care environment [24, 88, 134, 745, 837]. *Category IB*

V.B.3.b.ii. After gown removal, ensure that clothing and skin do not contact potentially contaminated environmental surfaces that could result in possible transfer of microorganism to other patients or environmental surfaces [72, 73]. *Category II*

V.B.4. Patient transport

V.B.4.a. In *acute care hospitals and long-term care and other residential settings*, limit transport and movement of patients outside of the room to medically-necessary purposes. *Category II*

V.B.4.b. When transport or movement in any healthcare setting is necessary, ensure that infected or colonized areas of the patient's body are contained and covered. *Category II*

V.B.4.c. Remove and dispose of contaminated PPE and perform hand hygiene prior to transporting patients on Contact Precautions. *Category II*

V.B.4.d. Don clean PPE to handle the patient at the transport destination. *Category II*

V.B.5. Patient-care equipment and instruments/devices

V.B.5.a. Handle patient-care equipment and instruments/devices according to Standard Precautions [739, 836]. *Category IB/IC*

V.B.5.b. In *acute care hospitals and long-term care and other residential settings*, use disposable noncritical patient-care equipment (e.g., blood pressure cuffs) or implement patient-dedicated use of such equipment. If common use of equipment for multiple patients is unavoidable, clean and disinfect such equipment before use on another patient [24, 88, 796, 836, 837, 854, 1016]. *Category IB*

V.B.5.c. In *home care settings*

V.B.5.c.i. Limit the amount of non-disposable patient-care equipment brought into the home of patients on Contact Precautions. Whenever possible, leave patient-care equipment in the home until discharge from home care services. *Category II*

V.B.5.c.ii. If noncritical patient-care equipment (e.g., stethoscope) cannot remain in the home, clean and disinfect items before taking them from the home using a low- to intermediate-level disinfectant. Alternatively, place contaminated reusable items in a plastic bag for transport and subsequent cleaning and disinfection. *Category II*

V.B.5.d. In *ambulatory settings*, place contaminated reusable noncritical patient-care equipment in a plastic bag for transport to a soiled utility area for reprocessing. *Category II*

V.B.6. Environmental measures

Ensure that rooms of patients on Contact Precautions are prioritized for frequent cleaning and disinfection (e.g., at least daily) with a focus on frequently-touched surfaces (e.g., bed rails, overbed table, bedside commode, lavatory surfaces in patient bathrooms, doorknobs) and equipment in the immediate vicinity of the patient [11, 24, 88, 746, 837]. *Category IB*

V.B.7. Discontinue Contact Precautions after signs and symptoms of the infection have resolved or according to pathogen-specific recommendations in Appendix A. *Category IB*

V.C. Droplet Precautions

V.C.1. Use Droplet Precautions as recommended in Appendix A for patients known or suspected to be infected with pathogens transmitted by respiratory droplets (i.e., large-particle droplets >5μ in size) that are generated by a patient who is coughing, sneezing or talking [14, 23, Steinberg, 1969 #1708, 41, 95, 103, 111, 112, 755, 756, 989, 1017]. *Category IB*

V.C.2. Patient placement

V.C.2.a. In acute care hospitals, place patients who require Droplet Precautions in a single-patient room when available *Category II* When single-patient rooms are in short supply, apply the following principles for making decisions on patient placement:

- Prioritize patients who have excessive cough and sputum production for single-patient room placement *Category II*
- Place together in the same room (cohort) patients who are infected the same pathogen and are suitable roommates [814-816]. *Category IB*
- If it becomes necessary to place patients who require Droplet Precautions in a room with a patient who does not have the same infection:
- Avoid placing patients on Droplet Precautions in the same room with patients who have conditions that may increase

the risk of adverse outcome from infection or that may facilitate transmission (e.g., those who are immunocompromised, have or have anticipated prolonged lengths of stay). *Category II*

- Ensure that patients are physically separated (i.e., >3 feet apart) from each other. Draw the privacy curtain between beds to minimize opportunities for close contact [103, 104 410]. *Category IB*
- Change protective attire and perform hand hygiene between contact with patients in the same room, regardless of whether one patient or both patients are on Droplet Precautions [741-743, 988, 1014, 1015]. *Category IB*

V.C.2.b. In *long-term care and other residential settings*, make decisions regarding patient placement on a case-by-case basis after considering infection risks to other patients in the room and available alternatives [410]. *Category II*

V.C.2.c. In *ambulatory settings*, place patients who require Droplet Precautions in an examination room or cubicle as soon as possible. Instruct patients to follow recommendations for Respiratory Hygiene/Cough Etiquette [447, 448 9, 828]. *Category II*

V.C.3. Use of personal protective equipment

V.C.3.a. Don a mask upon entry into the patient room or cubicle [14, 23, 41, 103, 111, 113, 115, 827]. *Category IB*

V.C.3.b. No recommendation for routinely wearing eye protection (e.g., goggle or face shield), in addition to a mask, for close contact with patients who require Droplet Precautions. *Unresolved issue*

V.C.3.c. For patients with suspected or proven SARS, avian influenza or pandemic influenza, refer to the following websites for the most current recommendations (www.**cdc**.gov/ncidod/**sars**/ ; www.**cdc**.gov/flu/**avian**/ ;www.**pandemicflu**.**gov**/) [134, 1018, 1019]

V.C.4. Patient transport

V.C.4.a. In *acute care hospitals and long-term care and other residential settings*, limit transport and movement of patients outside of the room to medically-necessary purposes. *Category II*

V.C.4.b. If transport or movement in any healthcare setting is necessary, instruct patient to wear a mask and follow Respiratory Hygiene/Cough Etiquette www.cdc.gov/flu/professionals/infectioncontrol/resphygiene.htm) . *Category IB*

V.C.4.c. No mask is required for persons transporting patients on Droplet Precautions. *Category II*

V.C.4.d. Discontinue Droplet Precautions after signs and symptoms have resolved or according to pathogen-specific recommendations in Appendix A. *Category IB*

V.D. Airborne Precautions

V.D.1. Use Airborne Precautions as recommended in Appendix A for patients known or suspected to be infected with infectious agents transmitted person-to-person by the airborne route (e.g., *M tuberculosis* [12], measles [34, 122, 1020], chickenpox [123, 773, 1021], disseminated herpes zoster [1022]. *Category IA/IC*

V.D.2. Patient placement

V.D.2.a. In *acute care hospitals and long-term care settings*, place patients who require Airborne Precautions in an AIIR that has been constructed in accordance with current guidelines [11-13]. *Category IA/IC*

V.D.2.a.i. Provide at least six (existing facility) or 12 (new construction/renovation) air changes per hour.

V.D.2.a.ii. Direct exhaust of air to the outside. If it is not possible to exhaust air from an AIIR directly to the outside, the air may be returned to the air-handling system or adjacent spaces if all air is directed through HEPA filters.

V.D.2.a.iii. Whenever an AIIR is in use for a patient on Airborne Precautions, monitor air pressure daily with visual indicators (e.g., smoke tubes, flutter strips), regardless of the presence of differential pressure sensing devices (e.g., manometers) [11, 12, 1023, 1024].

V.D.2.a.iv. Keep the AIIR door closed when not required for entry and exit.

V.D.2.b. When an AIIR is not available, transfer the patient to a facility that has an available AIIR [12]. *Category II*

V.D.2.c. In the event of an outbreak or exposure involving large numbers of patients who require Airborne Precautions:

- Consult infection control professionals before patient placement to determine the safety of alternative room that do not meet engineering requirements for an AIIR.
- Place together (cohort) patients who are presumed to have the same infection(based on clinical presentation and diagnosis when known) in areas of the facility that are away from other patients, especially patients who are at increased risk for infection (e.g., immunocompromised patients).
- Use temporary portable solutions (e.g., exhaust fan) to create a negative pressure environment in the converted area of the facility. Discharge air directly to the outside,away from people and air intakes, or direct all the air through HEPA filters before it is introduced to other air spaces [12] Category II

V.D.2.d. In *ambulatory settings*:

V.D.2.d.i. Develop systems (e.g., triage, signage) to identify patients with known or suspected infections that require Airborne Precautions upon entry into ambulatory settings [9, 12, 34, 127, 134]. *Category IA*

V.D.2.d.ii. Place the patient in an AIIR as soon as possible. If an AIIR is not available, place a surgical mask on the patient and place him/her in an examination room. Once the patient leaves, the room should remain vacant for the appropriate time, generally one hour, to allow for a full exchange of air [11, 12, 122]. *Category IB/IC*

V.D.2.d.iii. Instruct patients with a known or suspected airborne infection to wear a surgical mask and observe Respiratory Hygiene/Cough Etiquette. Once in an AIIR, the mask may be removed; the mask should remain on if the patient is not in an AIIR [12, 107, 145, 899]. *Category IB/IC*

V.D.3. Personnel restrictions

Restrict susceptible healthcare personnel from entering the rooms of patients known or suspected to have measles (rubeola), varicella (chickenpox), disseminated zoster, or smallpox if other immune healthcare personnel are available [17, 775]. *Category IB*

V.D.4. Use of PPE

V.D.4.a. Wear a fit-tested NIOSH-approved N95 or higher level respirator for respiratory protection when entering the room or home of a patient when the following diseases are suspected or confirmed:

- Infectious pulmonary or laryngeal tuberculosis or when infectious tuberculosis skin lesions are present and procedures that would aerosolize viable organisms (e.g., irrigation, incision and drainage, whirlpool treatments) are performed [12, 1025, 1026]. *Category IB*
- Smallpox (vaccinated and unvaccinated). Respiratory protection is recommended for all healthcare personnel, including those with a documented “take” after smallpox vaccination due to the risk of a genetically engineered virus against which the vaccine may not provide protection, or of exposure to a very large viral load (e.g., from high-risk aerosol-generating procedures, immunocompromised patients, hemorrhagic or flat smallpox [108, 129]. *Category II*

V.D.4.b. No recommendation is made regarding the use of PPE by healthcare personnel who are presumed to be immune to measles (rubeola) or varicella-zoster based on history of disease, vaccine, or serologic testing when caring for an individual with known or suspected measles, chickenpox or disseminated zoster, due to difficulties in establishing definite immunity [1027, 1028]. *Unresolved issue*

V.D.4.c. No recommendation is made regarding the type of personal protective equipment (i.e., surgical mask or respiratory protection with a N95 or higher respirator) to be worn by susceptible healthcare personnel who must have contact with patients with known or suspected measles, chickenpox or disseminated herpes zoster. *Unresolved issue*

V.D.5. Patient transport

V.D.5.a. In *acute care hospitals and long-term care and other residential settings*, limit transport and movement of patients outside of the room to medically-necessary purposes. *Category II*

V.D.5.b. If transport or movement outside an AIIR is necessary, instruct patients to wear a surgical mask, if possible, and observe Respiratory Hygiene/Cough Etiquette [12]. *Category II*

V.D.5.c. For patients with skin lesions associated with varicella or smallpox or draining skin lesions caused by *M. tuberculosis*, cover the affected areas to prevent aerosolization or contact with the infectious agent in skin lesions [108, 1025, 1026, 1029-1031]. *Category IB*

V.D.5.d. Healthcare personnel transporting patients who are on Airborne Precautions do not need to wear a mask or respirator during transport if the patient is wearing a mask and infectious skin lesions are covered. *Category II*

V.D.6. Exposure management

Immunize or provide the appropriate immune globulin to susceptible persons as soon as possible following unprotected contact (i.e., exposed) to a patient with measles, varicella or smallpox: *Category IA*

- Administer measles vaccine to exposed susceptible persons within 72 hours after the exposure or administer immune globulin within six days of the exposure event for high-risk persons in whom vaccine is contraindicated [17, 1032-1035].
- Administer varicella vaccine to exposed susceptible persons within 120 hours after the exposure or administer varicella immune globulin (VZIG or alternative product), when available, within 96 hours for high-risk persons in whom vaccine is contraindicated (e.g., immunocompromised patients, pregnant women, newborns whose mother's varicella onset was <5 days before or within 48 hours after delivery [888, 1035-1037]).
- Administer smallpox vaccine to exposed susceptible persons within 4 days after exposure [108, 1038-1040].

V.D.7. Discontinue Airborne Precautions according to pathogen-specific recommendations in Appendix A. *Category IB*

V.D.8. Consult CDC's "Guidelines for Preventing the Transmission of *Mycobacterium tuberculosis* in Health-Care Settings, 2005" [12] and the "Guideline for Environmental Infection Control in Health-Care Facilities" [11] for additional guidance on environment strategies for preventing transmission of tuberculosis in healthcare settings. The environmental recommendations in these guidelines may be applied to patients with other infections that require Airborne Precautions.

VI. Protective Environment (Table 4)

VI.A. Place allogeneic hematopoietic stem cell transplant (HSCT) patients in a Protective Environment as described in the "Guideline to Prevent Opportunistic Infections in HSCT Patients" [15], the "Guideline for Environmental Infection Control in Health-Care Facilities" [11], and the "Guidelines for Preventing Health-Care-Associated Pneumonia, 2003" [14] to reduce exposure to environmental fungi (e.g., *Aspergillus* sp) [157, 158]. *Category IB*

VI.B. No recommendation for placing patients with other medical conditions that are associated with increased risk for environmental fungal infections (e.g., aspergillosis) in a Protective Environment [11]. *Unresolved issue*

VI.C. For patients who require a Protective Environment, implement the following (see Table 5) [11, 15]

VI.C.1. Environmental controls

VI.C.1.a. Filtered incoming air using central or point-of-use high efficiency particulate (HEPA) filters capable of removing 99.97% of particles ≥0.3 µm in diameter [13]. *Category IB*

VI.C.1.b. Directed room airflow with the air supply on one side of the room that moves air across the patient bed and out through an exhaust on the opposite side of the room [13]. *Category IB*

VI.C.1.c. Positive air pressure in room relative to the corridor (pressure differential of ≥12.5 Pa [0.01-in water gauge]) [13]. *Category IB*

VI.C.1.c.i. Monitor air pressure daily with visual indicators (e.g., smoke tubes, flutter strips) [11, 1024]. *Category IA*

VI.C.1.d. Well-sealed rooms that prevent infiltration of outside air [13]. *Category IB*

VI.C.1.e. At least 12 air changes per hour [13]. *Category IB*

VI.C.2. Lower dust levels by using smooth, nonporous surfaces and finishes that can be scrubbed, rather than textured material (e.g., upholstery). Wet dust horizontal surfaces whenever dust detected and routinely clean crevices and sprinkler heads where dust may accumulate [940, 941]. *Category II*

VI.C.3. Avoid carpeting in hallways and patient rooms in areas [941]. *Category IB*

VI.C.4. Prohibit dried and fresh flowers and potted plants [942-944]. *Category II*

VI.D. Minimize the length of time that patients who require a Protective Environment are outside their rooms for diagnostic procedures and other activities [11, 158, 945]. *Category IB*

VI.E. During periods of construction, to prevent inhalation of respirable particles that could contain infectious spores, provide respiratory protection (e.g., N95 respirator) to patients who are medically fit to tolerate a respirator when they are required to leave the Protective Environment [945 158]. *Category II*

VI.E.1.a. No recommendation for fit-testing of patients who are using respirators. *Unresolved issue*

VI.E.1.b. No recommendation for use of particulate respirators when leaving the Protective Environment in the absence of construction. *Unresolved issue*

VI.F. Use of Standard and Transmission-Based Precautions in a Protective Environment.

VI.F.1. Use Standard Precautions as recommended for all patient interactions. *Category IA*

VI.F.2. Implement Droplet and Contact Precautions as recommended for diseases listed in Appendix A. Transmission-Based precautions for viral infections may need to be prolonged because of the patient's immunocompromised state and prolonged shedding of viruses [930] [1010] [928, 932] [1011]. *Category IB*

VI.F.3. Barrier precautions, (e.g., masks, gowns, gloves) are not required for healthcare personnel in the absence of suspected or confirmed infection in the patient or if they are not indicated according to Standard Precautions [15]. *Category II*

VI.F.4. Implement Airborne Precautions for patients who require a Protective Environment room and who also have an airborne infectious disease (e.g., pulmonary or laryngeal tuberculosis, acute varicella-zoster). *Category IA*

VI.F.4.a. Ensure that the Protective Environment is designed to maintain positive pressure [13]. *Category IB*

VI.F.4.b. Use an anteroom to further support the appropriate air-balance relative to the corridor and the Protective Environment; provide independent exhaust of contaminated air to the outside or place a HEPA filter in the exhaust duct if the return air must be recirculated [13, 1041]. *Category IB*

VI.F.4.c. If an anteroom is not available, place the patient in an AIIR and use portable, industrial-grade HEPA filters in the room to enhance filtration of spores [1042]. *Category II*

Appendix A:

Preamble The mode(s) and risk of transmission for each specific disease agent included in Appendix A were reviewed. Principle sources consulted for the development of disease-specific recommendations for Appendix A included infectious disease manuals and textbooks [833, 1043, 1044]. The published literature was searched for evidence of person-to-person transmission in healthcare and non-healthcare settings with a focus on reported outbreaks that would assist in developing recommendations for all settings where healthcare is delivered. Criteria used to assign Transmission-Based Precautions categories follow:

- A Transmission-Based Precautions category was assigned if there was strong evidence for person-to-person transmission via droplet, contact, or airborne routes in healthcare or non-healthcare settings and/or if patient factors (e.g., diapered infants, diarrhea, draining wounds) increased the risk of transmission
- Transmission-Based Precautions category assignments reflect the predominant mode(s) of transmission
- If there was no evidence for person-to-person transmission by droplet, contact or airborne routes, Standard Precautions were assigned
- If there was a low risk for person-to-person transmission and no evidence of healthcare-associated transmission, Standard Precautions were assigned
- Standard Precautions were assigned for bloodborne pathogens (e.g., hepatitis B and C viruses, human immunodeficiency virus) as per CDC recommendations for Universal Precautions issued in 1988 [780]. Subsequent experience has confirmed the efficacy of Standard Precautions to prevent exposure to infected blood and body fluid [778, 779, 866].

Additional information relevant to use of precautions was added in the comments column to assist the caregiver in decision-making. Citations were added as needed to support a change in or provide additional evidence for recommendations for a specific disease and for new infectious agents (e.g., SARS-CoV, avian influenza) that have been added to Appendix A. The reader may refer to more detailed discussion concerning modes of transmission and emerging pathogens in the background text and for MDRO control in Appendix B.

APPENDIX A[1]

TYPE AND DURATION OF PRECAUTIONS RECOMMENDED FOR SELECTED INFECTIONS AND CONDITIONS

Infection/Condition	Precautions		
	Type *	Duration †	Comments
Abscess			
Draining, major	C	DI	No dressing or containment of drainage; until drainage stops or can be contained by dressing
Draining, minor or limited	S		Dressing covers and contains drainage
Acquired human immunodeficiency syndrome (HIV)	S		Post-exposure chemoprophylaxis for some blood exposures [866].
Actinomycosis	S		Not transmitted from person to person
Adenovirus infection (see agent-specific guidance under gastroenteritis, conjuctivitis, pneumonia)			
Amebiasis	S		Person to person transmission is rare. Transmission in settings for the mentally challenged and in a family group has been reported [1045]. Use care when handling diapered infants and mentally challenged persons [1046].
Anthrax	S		Infected patients do not generally pose a transmission risk.
Cutaneous	S		Transmission through non-intact skin contact with draining lesions possible, therefore use Contact Precautions if large amount of uncontained drainage. Handwashing with soap and water preferable to use of waterless alcohol based antiseptics since alcohol does not

[1] Type of Precautions: A, Airborne Precautions; C, Contact; D, Droplet; S, Standard; when A, C, and D are specified, also use S.

† Duration of precautions: CN, until off antimicrobial treatment and culture-negative; DI, duration of illness (with wound lesions, DI means until wounds stop draining); DE, until environment completely decontaminated; U, until time specified in hours (hrs) after initiation of effective therapy; Unknown: criteria for establishing eradication of pathogen has not been determined

APPENDIX A[1]

TYPE AND DURATION OF PRECAUTIONS RECOMMENDED FOR SELECTED INFECTIONS AND CONDITIONS

Infection/Condition	Precautions		
	Type *	Duration †	Comments
			have sporicidal activity [983].
Pulmonary	S		Not transmitted from person to person
Environmental: aerosolizable spore-containing powder or other substance		DE	Until decontamination of environment complete [203] . Wear respirator (N95 mask or PAPRs), protective clothing; decontaminate persons with powder on them (http://www.cdc.gov/mmwr/preview/mmwrhtml/mm5135a3.htm) **Hand hygiene:** Handwashing for 30-60 seconds with soap and water or 2% chlorhexidene gluconate after spore contact (alcohol handrubs inactive against spores [983]. **Post-exposure prophylaxis following environmental exposure**: 60 days of antimicrobials (either doxycycline, ciprofloxacin, or levofloxacin) and post-exposure vaccine under IND
Antibiotic-associated colitis (see *Clostridium difficile*)			
Arthropod-borne viral encephalitides (eastern, western, Venezuelan equine encephalomyelitis; St Louis, California encephalitis; West Nile Virus) and viral fevers (dengue, yellow fever, Colorado tick fever)	S		Not transmitted from person to person except rarely by transfusion, and for West Nile virus by organ transplant, breastmilk or transplacentally [530, 1047]. Install screens in windows and doors in endemic areas Use DEET-containing mosquito repellants and clothing to cover extremities
Ascariasis	S		Not transmitted from person to person
Aspergillosis	S		Contact Precautions and Airborne Precautions if massive soft tissue infection with copious drainage and repeated irrigations required [154].
Avian influenza (see influenza, avian below)			
Babesiosis	S		Not transmitted from person to person except rarely by transfusion,
Blastomycosis, North American, cutaneous or pulmonary	S		Not transmitted from person to person
Botulism	S		Not transmitted from person to person
Bronchiolitis (see respiratory infections in infants and young children)	C	DI	Use mask according to Standard Precautions.

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TYPE AND DURATION OF PRECAUTIONS RECOMMENDED FOR SELECTED INFECTIONS AND CONDITIONS

Infection/Condition	Precautions		
	Type *	Duration †	Comments
Brucellosis (undulant, Malta, Mediterranean fever)	S		Not transmitted from person to person except rarely via banked spermatozoa and sexual contact [1048, 1049]. Provid antimicrobial prophylaxis following laboratory exposure [1050].
Campylobacter gastroenteritis (see gastroenteritis)			
Candidiasis, all forms including mucocutaneous	S		
Cat-scratch fever (benign inoculation lymphoreticulosis)	S		Not transmitted from person to person
Cellulitis	S		
Chancroid (soft chancre) (*H. ducreyi*)	S		Transmitted sexually from person to person
Chickenpox (see varicella)			
Chlamydia trachomatis			
Conjunctivitis	S		
Genital (lymphogranuloma venereum)	S		
Pneumonia (infants ≤ 3 mos. of age))	S		
Chlamydia pneumoniae	S		Outbreaks in institutionalized populations reported, rarely [1051, 1052]
Cholera (see gastroenteritis)			
Closed-cavity infection			
Open drain in place; limited or minor drainage	S		Contact Precautions if there is copious uncontained drainage
No drain or closed drainage system in place	S		
Clostridium			
C. botulinum	S		Not transmitted from person to person
C. difficile (see Gastroenteritis, *C. difficile)*	C	DI	
C. perfringens			
Food poisoning	S		Not transmitted from person to person
Gas gangrene	S		Transmission from person to person rare; one outbreak in a surgical setting reported [1053]. Use Contact Precautions if wound drainage is

APPENDIX A[1]

TYPE AND DURATION OF PRECAUTIONS RECOMMENDED FOR SELECTED INFECTIONS AND CONDITIONS

Infection/Condition	Precautions		
	Type *	Duration †	Comments
			extensive.
Coccidioidomycosis (valley fever)			
Draining lesions	S		Not transmitted from person to person except under extraordinary circumstances because the infectious arthroconidial form of *Coccidioides immitis* is not produced in humans [1054] .
Pneumonia	S		Not transmitted from person to person except under extraordinary circumstances, (e.g., inhalation of aerosolized tissue phase endospores during necropsy, transplantation of infected lung) because the infectious arthroconidial form of *Coccidioides immitis* is not produced in humans [1054, 1055].
Colorado tick fever	S		Not transmitted from person to person
Congenital rubella	C	Until 1 yr of age	Standard Precautions if nasopharyngeal and urine cultures repeatedly neg. after 3 mos. of age
Conjunctivitis			
Acute bacterial	S		
Chlamydia	S		
Gonococcal	S		
Acute viral (acute hemorrhagic)	C	DI	Adenovirus most common; enterovirus 70 [1056], Coxsackie virus A24 [1057]) also associated with community outbreaks. Highly contagious; outbreaks in eye clinics, pediatric and neonatal settings, institutional settings reported. Eye clinics should follow Standard Precautions when handling patients with conjunctivitis. Routine use of infection control measures in the handling of instruments and equipment will prevent the occurrence of outbreaks in this and other settings. [460, 814, 1058, 1059 461, 1060] .
Corona virus associated with SARS (SARS-CoV) (see severe acute respiratory syndrome)			

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TYPE AND DURATION OF PRECAUTIONS RECOMMENDED FOR SELECTED INFECTIONS AND CONDITIONS

Infection/Condition	Precautions		
	Type *	Duration †	Comments
Coxsackie virus disease (see enteroviral infection)			
Creutzfeldt-Jakob disease CJD, vCJD	S		Use disposable instruments or special sterilization/disinfection for surfaces, objects contaminated with neural tissue if CJD or vCJD suspected and has not been R/O; No special burial procedures [1061]
Croup (see respiratory infections in infants and young children)			
Crimean-Congo Fever (see Viral Hemorrhagic Fever)	S		
Cryptococcosis	S		Not transmitted from person to person, except rarely via tissue and corneal transplant [1062, 1063]
Cryptosporidiosis (see gastroenteritis)			
Cysticercosis	S		Not transmitted from person to person
Cytomegalovirus infection, including in neonates and immunosuppressed patients	S		No additional precautions for pregnant HCWs
Decubitus ulcer (see Pressure ulcer)			
Dengue fever	S		Not transmitted from person to person
Diarrhea, acute-infective etiology suspected (see gastroenteritis)			
Diphtheria			
Cutaneous	C	CN	Until 2 cultures taken 24 hrs. apart negative
Pharyngeal	D	CN	Until 2 cultures taken 24 hrs. apart negative
Ebola virus (see viral hemorrhagic fevers)			
Echinococcosis (hydatidosis)	S		Not transmitted from person to person
Echovirus (see enteroviral infection)			
Encephalitis or encephalomyelitis (see specific etiologic agents)			
Endometritis (endomyometritis)	S		
Enterobiasis (pinworm disease, oxyuriasis)	S		
Enterococcus species (see multidrug-resistant organisms if			

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TYPE AND DURATION OF PRECAUTIONS RECOMMENDED FOR SELECTED INFECTIONS AND CONDITIONS

Infection/Condition	Precautions		
	Type *	Duration †	Comments
epidemiologically significant or vancomycin resistant)			
Enterocolitis, *C. difficile* (see *C. difficile,* gastroenteritis)			
Enteroviral infections (i.e., Group A and B Coxsackie viruses and Echo viruses) (excludes polio virus)	S		Use Contact Precautions for diapered or incontinent children for duration of illness and to control institutional outbreaks
Epiglottitis, due to *Haemophilus influenzae* type b	D	U 24 hrs	See specific disease agents for epiglottitis due to other etiologies)
Epstein-Barr virus infection, including infectious mononucleosis	S		
Erythema infectiosum (also see Parvovirus B19)			
Escherichia coli gastroenteritis (see gastroenteritis)			
Food poisoning			
Botulism	S		Not transmitted from person to person
C. perfringens or welchii	S		Not transmitted from person to person
Staphylococcal	S		Not transmitted from person to person
Furunculosis, staphylococcal	S		Contact if drainage not controlled. Follow institutional policies if MRSA
Infants and young children	C	DI	
Gangrene (gas gangrene)	S		Not transmitted from person to person
Gastroenteritis	S		Use Contact Precautions for diapered or incontinent persons for the duration of illness or to control institutional outbreaks for gastroenteritis caused by all of the agents below
Adenovirus	S		Use Contact Precautions for diapered or incontinent persons for the duration of illness or to control institutional outbreaks
Campylobacter species	S		Use Contact Precautions for diapered or incontinent persons for the duration of illness or to control institutional outbreaks
Cholera *(Vibrio cholerae)*	S		Use Contact Precautions for diapered or incontinent persons for the duration of illness or to control institutional outbreaks
C. difficile	C	DI	Discontinue antibiotics if appropriate. Do not share electronic thermometers [853, 854]; ensure consistent environmental cleaning and

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TYPE AND DURATION OF PRECAUTIONS RECOMMENDED FOR SELECTED INFECTIONS AND CONDITIONS

Infection/Condition	Precautions		
	Type *	Duration †	Comments
			disinfection. Hypochlorite solutions may be required for cleaning if transmission continues [847]. Handwashing with soap and water preferred because of the absence of sporicidal activity of alcohol in waterless antiseptic handrubs [983].
Cryptosporidium species	S		Use Contact Precautions for diapered or incontinent persons for the duration of illness or to control institutional outbreaks
E. coli			
Enteropathogenic O157:H7 and other shiga toxin-producing Strains	S		Use Contact Precautions for diapered or incontinent persons for the duration of illness or to control institutional outbreaks
Other species	S		Use Contact Precautions for diapered or incontinent persons for the duration of illness or to control institutional outbreaks
Giardia lamblia	S		Use Contact Precautions for diapered or incontinent persons for the duration of illness or to control institutional outbreaks
Noroviruses	S		Use Contact Precautions for diapered or incontinent persons for the duration of illness or to control institutional outbreaks. Persons who clean areas heavily contaminated with feces or vomitus may benefit from wearing masks since virus can be aerosolized from these body substances [142, 147 148]; ensure consistent environmental cleaning and disinfection with focus on restrooms even when apparently unsoiled [273, 1064]). Hypochlorite solutions may be required when there is continued transmission [290-292]. Alcohol is less active, but there is no evidence that alcohol antiseptic handrubs are not effective for hand decontamination [294]. Cohorting of affected patients to separate airspaces and toilet facilities may help interrupt transmission during outbreaks.
Rotavirus	C	DI	Ensure consistent environmental cleaning and disinfection and frequent removal of soiled diapers. Prolonged shedding may occur in

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TYPE AND DURATION OF PRECAUTIONS RECOMMENDED FOR SELECTED INFECTIONS AND CONDITIONS

Infection/Condition	Precautions		
	Type *	Duration †	Comments
			both immunocompetent and immunocompromised children and the elderly [932, 933].
Salmonella species (including *S. typhi*)	S		Use Contact Precautions for diapered or incontinent persons for the duration of illness or to control institutional outbreaks
Shigella species (Bacillary dysentery)	S		Use Contact Precautions for diapered or incontinent persons for the duration of illness or to control institutional outbreaks
Vibrio parahaemolyticus	S		Use Contact Precautions for diapered or incontinent persons for the duration of illness or to control institutional outbreaks
Viral (if not covered elsewhere)	S		Use Contact Precautions for diapered or incontinent persons for the duration of illness or to control institutional outbreaks
Yersinia enterocolitica	S		Use Contact Precautions for diapered or incontinent persons for the duration of illness or to control institutional outbreaks
German measles (see rubella; see congenital rubella)			
Giardiasis (see gastroenteritis)			
Gonococcal ophthalmia neonatorum (gonorrheal ophthalmia, acute conjunctivitis of newborn)	S		
Gonorrhea	S		
Granuloma inguinale (Donovanosis, granuloma venereum)	S		
Guillain-Barré' syndrome	S		Not an infectious condition
Haemophilus influenzae (see disease-specific recommendations)			
Hand, foot, and mouth disease (see enteroviral infection)			
Hansen's Disease (see Leprosy)			
Hantavirus pulmonary syndrome	S		Not transmitted from person to person
Helicobacter pylori	S		
Hepatitis, viral			
Type A	S		Provide hepatitis A vaccine post-exposure as recommended [1065]
Diapered or incontinent patients	C		Maintain Contact Precautions in infants and children <3 years of age

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TYPE AND DURATION OF PRECAUTIONS RECOMMENDED FOR SELECTED INFECTIONS AND CONDITIONS

Infection/Condition	Precautions		
	Type *	Duration †	Comments
			for duration of hospitalization; for children 3-14 yrs. of age for 2 weeks after onset of symptoms; >14 yrs. of age for 1 week after onset of symptoms [833, 1066, 1067].
Type B-HBsAg positive; acute or chronic	S		See specific recommendations for care of patients in hemodialysis centers [778]
Type C and other unspecified non-A, non-B	S		See specific recommendations for care of patients in hemodialysis centers [778]
Type D (seen only with hepatitis B)	S		
Type E	S		Use Contact Precautions for diapered or incontinent individuals for the duration of illness [1068]
Type G	S		
Herpangina (see enteroviral infection)			
Hookworm	S		
Herpes simplex (*Herpesvirus hominis*)			
Encephalitis	S		
Mucocutaneous, disseminated or primary, severe	C	Until lesions dry and crusted	
Mucocutaneous, recurrent (skin, oral, genital)	S		
Neonatal	C	Until lesions dry and crusted	Also, for asymptomatic, exposed infants delivered vaginally or by C-section and if mother has active infection and membranes have been ruptured for more than 4 to 6 hrs until infant surface cultures obtained at 24-36 hrs. of age negative after 48 hrs incubation [1069, 1070]
Herpes zoster (varicella-zoster) (shingles)			
Disseminated disease in any patient Localized disease in immunocompromised patient until disseminated infection ruled out	A,C	DI	Susceptible HCWs should not enter room if immune caregivers are available; no recommendation for protection of immune HCWs; no recommendation for type of protection, i.e. surgical mask or respirator; for susceptible HCWs.

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TYPE AND DURATION OF PRECAUTIONS RECOMMENDED FOR SELECTED INFECTIONS AND CONDITIONS

Infection/Condition	Precautions		
	Type *	Duration †	Comments
Localized in patient with intact immune system with lesions that can be contained/covered	S	DI	Susceptible HCWs should not provide direct patient care when other immune caregivers are available.
Histoplasmosis	S		Not transmitted from person to person
Human immunodeficiency virus (HIV)	S		Post-exposure chemoprophylaxis for some blood exposures [866].
Human metapneumovirus	C	DI	HAI reported [1071], but route of transmission not established [823]. Assumed to be Contact transmission as for RSV since the viruses are closely related and have similar clinical manifestations and epidemiology. Wear masks according to Standard Precautions..
Impetigo	C	U 24 hrs	
Infectious mononucleosis	S		
Influenza			
Human (seasonal influenza)	D	5 days except DI in immuno compromised persons	Single patient room when available or cohort; avoid placement with high-risk patients; mask patient when transported out of room; chemoprophylaxis/vaccine to control/prevent outbreaks [611]. Use gown and gloves according to Standard Precautions may be especially important in pediatric settings. Duration of precautions for immunocompromised patients cannot be defined; prolonged duration of viral shedding (i.e. for several weeks) has been observed; implications for transmission are unknown [930].
Avian (e.g., H5N1, H7, H9 strains))			See www.cdc.gov/flu/avian/professional/infect-control.htm for current avian influenza guidance.
Pandemic influenza (also a human influenza virus)	D	5 days from onset of symptoms	See http://www.pandemicflu.gov for current pandemic influenza guidance.
Kawasaki syndrome	S		Not an infectious condition
Lassa fever (see viral hemorrhagic fevers)			

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TYPE AND DURATION OF PRECAUTIONS RECOMMENDED FOR SELECTED INFECTIONS AND CONDITIONS

Infection/Condition	Precautions		
	Type *	Duration †	Comments
Legionnaires' disease	S		Not transmitted from person to person
Leprosy	S		
Leptospirosis	S		Not transmitted from person to person
Lice			http://www.cdc.gov/ncidod/dpd/parasites/lice/default.htm
Head (pediculosis)	C	U 24 hrs	
Body	S		Transmitted person to person through infested clothing. Wear gown and gloves when removing clothing; bag and wash clothes according to CDC guidance above
Pubic	S		Transmitted person to person through sexual contact
Listeriosis (listeria monocytogenes)	S		Person-to-person transmission rare; cross-transmission in neonatal settings reported [1072, 1073 1074, 1075]
Lyme disease	S		Not transmitted from person to person
Lymphocytic choriomeningitis	S		Not transmitted from person to person
Lymphogranuloma venereum	S		
Malaria	S		Not transmitted from person to person except through transfusion rarely and through a failure to follow Standard Precautions during patient care [1076-1079]. Install screens in windows and doors in endemic areas. Use DEET-containing mosquito repellants and clothing to cover extremities
Marburg virus disease (see viral hemorrhagic fevers)			
Measles (rubeola)	A	4 days after onset of rash; DI in immune compromised	Susceptible HCWs should not enter room if immune care providers are available; no recommendation for face protection for immune HCW; no recommendation for type of face protection for susceptible HCWs, i.e., mask or respirator [1027, 1028]. For exposed susceptibles, post-exposure vaccine within 72 hrs. or immune globulin within 6 days when available [17, 1032, 1034]. Place exposed susceptible patients on Airborne Precautions and exclude susceptible healthcare personnel

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TYPE AND DURATION OF PRECAUTIONS RECOMMENDED FOR SELECTED INFECTIONS AND CONDITIONS

Infection/Condition	Precautions		
	Type *	Duration †	Comments
			from duty from day 5 after first exposure to day 21 after last exposure, regardless of post-exposure vaccine [17].
Melioidosis, all forms	S		Not transmitted from person to person
Meningitis			
Aseptic (nonbacterial or viral; also see enteroviral infections)	S		Contact for infants and young children
Bacterial, gram-negative enteric, in neonates	S		
Fungal	S		
Haemophilus influenzae, type b known or suspected	D	U 24 hrs	
Listeria monocytogenes (See Listeriosis)	S		
Neisseria meningitidis (meningococcal) known or suspected	D	U 24 hrs	See meningococcal disease below
Streptococcus pneumoniae	S		
M. tuberculosis	S		Concurrent, active pulmonary disease or draining cutaneous lesions may necessitate addition of Contact and/or Airborne Precautions; For children, airborne precautions until active tuberculosis ruled out in visiting family members (see tuberculosis below) [42]
Other diagnosed bacterial	S		
Meningococcal disease: sepsis, pneumonia, meningitis	D	U 24 hrs	Postexposure chemoprophylaxis for household contacts, HCWs exposed to respiratory secretions; postexposure vaccine only to control outbreaks [15, 17].
Molluscum contagiosum	S		
Monkeypox	A,C	A-Until monkeypox confirmed and smallpox excluded C-Until lesions crusted	Use See www.cdc.gov/ncidod/monkeypox for most current recommendations. Transmission in hospital settings unlikely [269]. Pre- and post-exposure smallpox vaccine recommended for exposed HCWs

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TYPE AND DURATION OF PRECAUTIONS RECOMMENDED FOR SELECTED INFECTIONS AND CONDITIONS

Infection/Condition	Precautions		
	Type *	Duration †	Comments
Mucormycosis	S		
Multidrug-resistant organisms (MDROs), infection or colonization (e.g., MRSA, VRE, VISA/VRSA, ESBLs, resistant *S. pneumoniae*)	S/C		MDROs judged by the infection control program, based on local, state, regional, or national recommendations, to be of clinical and epidemiologic significance. Contact Precautions recommended in settings with evidence of ongoing transmission, acute care settings with increased risk for transmission or wounds that cannot be contained by dressings. See recommendations for management options in Management of Multidrug-Resistant Organisms In Healthcare Settings, 2006 [870]. Contact state health department for guidance regarding new or emerging MDRO.
Mumps (infectious parotitis)	D	U 9 days	After onset of swelling; susceptible HCWs should not provide care if immune caregivers are available. Note: (Recent assessment of outbreaks in healthy 18-24 year olds has indicated that salivary viral shedding occurred early in the course of illness and that 5 days of isolation after onset of parotitis may be appropriate in community settings; however the implications for healthcare personnel and high-risk patient populations remain to be clarified.)
Mycobacteria, nontuberculosis (atypical)			Not transmitted person-to-person
Pulmonary	S		
Wound	S		
Mycoplasma pneumonia	D	DI	
Necrotizing enterocolitis	S		Contact Precautions when cases clustered temporally [1080-1083].
Nocardiosis, draining lesions, or other presentations	S		Not transmitted person-to-person
Norovirus (see gastroenteritis)			
Norwalk agent gastroenteritis (see gastroenteritis)			
Orf	S		

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TYPE AND DURATION OF PRECAUTIONS RECOMMENDED FOR SELECTED INFECTIONS AND CONDITIONS

Infection/Condition	Precautions		
	Type [*]	**Duration** [†]	**Comments**
Parainfluenza virus infection, respiratory in infants and young children	C	DI	Viral shedding may be prolonged in immunosuppressed patients [1009, 1010]. Reliability of antigen testing to determine when to remove patients with prolonged hospitalizations from Contact Precautions uncertain.
Parvovirus B19 (Erythema infectiosum)	D		Maintain precautions for duration of hospitalization when chronic disease occurs in an immunocompromised patient. For patients with transient aplastic crisis or red-cell crisis, maintain precautions for 7 days. Duration of precautions for immunosuppressed patients with persistently positive PCR not defined, but transmission has occurred [929].
Pediculosis (lice)	C	U 24 hrs after treatment	
Pertussis (whooping cough)	D	U 5 days	Single patient room preferred. Cohorting an option. Post-exposure chemoprophylaxis for household contacts and HCWs with prolonged exposure to respiratory secretions [863]. Recommendations for Tdap vaccine in adults under development.
Pinworm infection (Enterobiasis)	S		
Plague *(Yersinia pestis)*			
Bubonic	S		
Pneumonic	D	U 48 hrs	Antimicrobial prophylaxis for exposed HCW [207].
Pneumonia			
Adenovirus	D, C	DI	Outbreaks in pediatric and institutional settings reported [376, 1084-1086]. In immunocompromised hosts, extend duration of Droplet and Contact Precautions due to prolonged shedding of virus [931]
Bacterial not listed elsewhere (including gram-negative bacterial)	S		
B. cepacia in patients with CF, including respiratory tract colonization	C	Unknown	Avoid exposure to other persons with CF; private room preferred. Criteria for D/C precautions not established. See CF Foundation guideline [20]

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TYPE AND DURATION OF PRECAUTIONS RECOMMENDED FOR SELECTED INFECTIONS AND CONDITIONS

Infection/Condition	Precautions		
	Type *	Duration †	Comments
B. cepacia in patients without CF(see Multidrug-resistant organisms)			
Chlamydia	S		
Fungal	S		
Haemophilus influenzae, type b			
Adults	S		
Infants and children	D	U 24 hrs	
Legionella spp.	S		
Meningococcal	D	U 24 hrs	See meningococcal disease above
Multidrug-resistant bacterial (see multidrug-resistant organisms)			
Mycoplasma (primary atypical pneumonia)	D	DI	
Pneumococcal pneumonia	S		Use Droplet Precautions if evidence of transmission within a patient care unit or facility [196-198, 1087]
Pneumocystis jiroveci (*Pneumocystis carinii*)	S		Avoid placement in the same room with an immunocompromised patient.
Staphylococcus aureus	S		For MRSA, see MDROs
Streptococcus, group A			
Adults	D	U 24 hrs	See streptococcal disease (group A streptococcus) below Contact precautions if skin lesions present
Infants and young children	D	U 24 hrs	Contact Precautions if skin lesions present
Varicella-zoster (See Varicella-Zoster)			
Viral			
Adults	S		
Infants and young children (see respiratory infectious disease, acute, or specific viral agent)			
Poliomyelitis	C	DI	
Pressure ulcer (decubitus ulcer, pressure sore) infected			

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TYPE AND DURATION OF PRECAUTIONS RECOMMENDED FOR SELECTED INFECTIONS AND CONDITIONS

Infection/Condition	Precautions		
	Type *	Duration †	Comments
Major	C	DI	If no dressing or containment of drainage; until drainage stops or can be contained by dressing
Minor or limited	S		If dressing covers and contains drainage
Prion disease (See Creutzfeld-Jacob Disease)			
Psittacosis (ornithosis) (*Chlamydia psittaci)*	S		Not transmitted from person to person
Q fever	S		
Rabies	S		Person to person transmission rare; transmission via corneal, tissue and organ transplants has been reported [539, 1088]. If patient has bitten another individual or saliva has contaminated an open wound or mucous membrane, wash exposed area thoroughly and administer postexposure prophylaxis. [1089]
Rat-bite fever (*Streptobacillus moniliformis* disease, *Spirillum minus* disease)	S		Not transmitted from person to person
Relapsing fever	S		Not transmitted from person to person
Resistant bacterial infection or colonization (see multidrug-resistant organisms)			
Respiratory infectious disease, acute (if not covered elsewhere)			
Adults	S		
Infants and young children	C	DI	Also see syndromes or conditions listed in Table 2
Respiratory syncytial virus infection, in infants, young children and immunocompromised adults	C	DI	Wear mask according to Standard Precautions [24] CB [116, 117]. In immunocompromised patients, extend the duration of Contact Precautions due to prolonged shedding [928]). Reliability of antigen testing to determine when to remove patients with prolonged hospitalizations from Contact Precautions uncertain.
Reye's syndrome	S		Not an infectious condition
Rheumatic fever	S		Not an infectious condition
Rhinovirus	D	DI	Droplet most important route of transmission [104, 1090]. Outbreaks have

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TYPE AND DURATION OF PRECAUTIONS RECOMMENDED FOR SELECTED INFECTIONS AND CONDITIONS

Infection/Condition	Precautions		
	Type *	Duration †	Comments
			occurred in NICUs and LTCFs [413, 1091, 1092]. Add Contact Precautions if copious moist secretions and close contact likely to occur (e.g., young infants) [111, 833].
Rickettsial fevers, tickborne (Rocky Mountain spotted fever, tickborne typhus fever)	S		Not transmitted from person to person except through transfusion, rarely
Rickettsialpox (vesicular rickettsiosis)	S		Not transmitted from person to person
Ringworm (dermatophytosis, dermatomycosis, tinea)	S		Rarely, outbreaks have occurred in healthcare settings, (e.g., NICU [1093], rehabilitation hospital [1094]. Use Contact Precautions for outbreak.
Ritter's disease (staphylococcal scalded skin syndrome)	C	DI	See staphylococcal disease, scalded skin syndrome below
Rocky Mountain spotted fever	S		Not transmitted from person to person except through transfusion, rarely
Roseola infantum (exanthem subitum; caused by HHV-6)	S		
Rotavirus infection (see gastroenteritis)			
Rubella (German measles) (also see congenital rubella)	D	U 7 days after onset of rash	Susceptible HCWs should not enter room if immune caregivers are available. No recommendation for wearing face protection (e.g., a surgical mask) if immune. Pregnant women who are not immune should not care for these patients [17, 33]. Administer vaccine within three days of exposure to non-pregnant susceptible individuals. Place exposed susceptible patients on Droplet Precautions; exclude susceptible healthcare personnel from duty from day 5 after first exposure to day 21 after last exposure, regardless of post-exposure vaccine.
Rubeola (see measles)			
Salmonellosis (see gastroenteritis)			
Scabies	C	U 24	
Scalded skin syndrome, staphylococcal	C	DI	See staphylococcal disease, scalded skin syndrome below)
Schistosomiasis (bilharziasis)	S		

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TYPE AND DURATION OF PRECAUTIONS RECOMMENDED FOR SELECTED INFECTIONS AND CONDITIONS

Infection/Condition	Precautions		
	Type *	**Duration** †	**Comments**
Severe acute respiratory syndrome (SARS)	A, D,C	DI plus 10 days after resolution of fever, provided respiratory symptoms are absent or improving	Airborne Precautions preferred; D if AIIR unavailable. N95 or higher respiratory protection; surgical mask if N95 unavailable; eye protection (goggles, face shield); aerosol-generating procedures and "supershedders" highest risk for transmission via small droplet nuclei and large droplets [93, 94, 96].Vigilant environmental disinfection (see www.cdc.gov/ncidod/sars)
Shigellosis (see gastroenteritis)			
Smallpox (variola; see vaccinia for management of vaccinated persons)	A,C	DI	Until all scabs have crusted and separated (3-4 weeks). Non-vaccinated HCWs should not provide care when immune HCWs are available; N95 or higher respiratory protection for susceptible and successfully vaccinated individuals; postexposure vaccine within 4 days of exposure protective [108, 129, 1038-1040].
Sporotrichosis	S		
Spirillum minor disease (rat-bite fever)	S		Not transmitted from person to person
Staphylococcal disease (*S aureus*)			
Skin, wound, or burn			
Major	C	DI	No dressing or dressing does not contain drainage adequately
Minor or limited	S		Dressing covers and contains drainage adequately
Enterocolitis	S		Use Contact Precautions for diapered or incontinent children for duration of illness
Multidrug-resistant (see multidrug-resistant organisms)			
Pneumonia	S		
Scalded skin syndrome	C	DI	Consider healthcare personnel as potential source of nursery, NICU outbreak [1095].
Toxic shock syndrome	S		
Streptobacillus moniliformis disease (rat-bite fever)	S		Not transmitted from person to person

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TYPE AND DURATION OF PRECAUTIONS RECOMMENDED FOR SELECTED INFECTIONS AND CONDITIONS

Infection/Condition	Precautions		
	Type *	Duration †	Comments
Streptococcal disease (group A streptococcus)			
Skin, wound, or burn			
Major	C,D	U 24 hrs	No dressing or dressing does not contain drainage adequately
Minor or limited	S		Dressing covers and contains drainage adequately
Endometritis (puerperal sepsis)	S		
Pharyngitis in infants and young children	D	U 24 hrs	
Pneumonia	D	U 24 hrs	
Scarlet fever in infants and young children	D	U 24 hrs	
Serious invasive disease	D	U24 hrs	Outbreaks of serious invasive disease have occurred secondary to transmission among patients and healthcare personnel [162, 972, 1096-1098] Contact Precautions for draining wound as above; follow rec. for antimicrobial prophylaxis in selected conditions [160].
Streptococcal disease (group B streptococcus), neonatal	S		
Streptococcal disease (not group A or B) unless covered elsewhere	S		
Multidrug-resistant (see multidrug-resistant organisms)			
Strongyloidiasis	S		
Syphilis			
Latent (tertiary) and seropositivity without lesions	S		
Skin and mucous membrane, including congenital, primary, Secondary	S		
Tapeworm disease			
Hymenolepis nana	S		Not transmitted from person to person
Taenia solium (pork)	S		
Other	S		
Tetanus	S		Not transmitted from person to person
Tinea (e.g., dermatophytosis, dermatomycosis, ringworm)	S		Rare episodes of person-to-person transmission

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TYPE AND DURATION OF PRECAUTIONS RECOMMENDED FOR SELECTED INFECTIONS AND CONDITIONS

Infection/Condition	Precautions		
	Type *	Duration †	Comments
Toxoplasmosis	S		Transmission from person to person is rare; vertical transmission from mother to child, transmission through organs and blood transfusion rare
Toxic shock syndrome (staphylococcal disease, streptococcal disease)	S		Droplet Precautions for the first 24 hours after implementation of antibiotic therapy if Group A streptococcus is a likely etiology
Trachoma, acute	S		
Transmissible spongiform encephalopathy (see Creutzfeld-Jacob disease, CJD, vCJD)			
Trench mouth (Vincent's angina)	S		
Trichinosis	S		
Trichomoniasis	S		
Trichuriasis (whipworm disease)	S		
Tuberculosis *(M. tuberculosis)*			
Extrapulmonary, draining lesion)	A,C		Discontinue precautions only when patient is improving clinically, and drainage has ceased or there are three consecutive negative cultures of continued drainage [1025, 1026]. Examine for evidence of active pulmonary tuberculosis.
Extrapulmonary, no draining lesion, meningitis	S		Examine for evidence of pulmonary tuberculosis. For infants and children, use Airborne Precautions until active pulmonary tuberculosis in visiting family members ruled out [42]
Pulmonary or laryngeal disease, confirmed	A		Discontinue precautions only when patient on effective therapy is improving clinically and has three consecutive sputum smears negative for acid-fast bacilli collected on separate days(MMWR 2005; 54: RR-17 http://www.cdc.gov/mmwr/preview/mmwrhtml/rr5417a1.htm?s_cid=rr5417a1_e) [12].
Pulmonary or laryngeal disease, suspected	A		Discontinue precautions only when the likelihood of infectious TB

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TYPE AND DURATION OF PRECAUTIONS RECOMMENDED FOR SELECTED INFECTIONS AND CONDITIONS

Infection/Condition	Precautions		
	Type *	Duration †	Comments
			disease is deemed negligible, and either 1) there is another diagnosis that explains the clinical syndrome or 2) the results of three sputum smears for AFB are negative. Each of the three sputum specimens should be collected 8-24 hours apart, and at least one should be an early morning specimen
Skin-test positive with no evidence of current active disease	S		
Tularemia			
Draining lesion	S		Not transmitted from person to person
Pulmonary	S		Not transmitted from person to person
Typhoid (*Salmonella typhi*) fever (see gastroenteritis)			
Typhus			
Rickettsia prowazekii (Epidemic or Louse-borne typhus)	S		Transmitted from person to person through close personal or clothing contact
Rickettsia typhi	S		Not transmitted from person to person
Urinary tract infection (including pyelonephritis), with or without urinary catheter	S		
Vaccinia (vaccination site, adverse events following vaccination) *			Only vaccinated HCWs have contact with active vaccination sites and care for persons with adverse vaccinia events; if unvaccinated, only HCWs without contraindications to vaccine may provide care.
Vaccination site care (including autoinoculated areas)	S		Vaccination recommended for vaccinators; for newly vaccinated HCWs: semi-permeable dressing over gauze until scab separates, with dressing change as fluid accumulates, ~3-5 days; gloves, hand hygiene for dressing change; vaccinated HCW or HCW without contraindication to vaccine for dressing changes [205, 221, 225].
Eczema vaccinatum	C	Until lesions dry and crusted, scabs separated	For contact with virus-containing lesions and exudative material
Fetal vaccinia	C		
Generalized vaccinia	C		

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TYPE AND DURATION OF PRECAUTIONS RECOMMENDED FOR SELECTED INFECTIONS AND CONDITIONS

Infection/Condition	Precautions		
	Type *	Duration †	Comments
Progressive vaccinia	C		
Postvaccinia encephalitis	S		
Blepharitis or conjunctivitis	S/C		Use Contact Precautions if there is copious drainage
Iritis or keratitis	S		
Vaccinia-associated erythema multiforme (Stevens Johnson Syndrome)	S		Not an infectious condition
Secondary bacterial infection (e.g., S. aureus, group A beta hemolytic streptococcus	S/C		Follow organism-specific (strep, staph most frequent) recommendations and consider magnitude of drainage
Varicella Zoster	A,C	Until lesions dry and crusted	Susceptible HCWs should not enter room if immune caregivers are available; no recommendation for face protection of immune HCWs; no recommendation for type of protection, i.e. surgical mask or respirator for susceptible HCWs. In immunocompromised host with varicella pneumonia, prolong duration of precautions for duration of illness. Post-exposure prophylaxis: provide post-exposure vaccine ASAP but within 120 hours; for susceptible exposed persons for whom vaccine is contraindicated (immunocompromised persons, pregnant women, newborns whose mother's varicella onset is $\leq$5days before delivery or within 48 hrs after delivery) provide VZIG, when available, within 96 hours; if unavailable, use IVIG, Use Airborne Precautions for exposed susceptible persons and exclude exposed susceptible healthcare workers beginning 8 days after first exposure until 21 days after last exposure or 28 if received VZIG, regardless of postexposure vaccination. [1036].
Variola (see smallpox)			
Vibrio parahaemolyticus (see gastroenteritis)			
Vincent's angina (trench mouth)	S		
Viral hemorrhagic fevers	S, D, C	DI	Single-patient room preferred. Emphasize: 1) use of sharps safety

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TYPE AND DURATION OF PRECAUTIONS RECOMMENDED FOR SELECTED INFECTIONS AND CONDITIONS

Infection/Condition	Precautions		
	Type *	Duration †	Comments
due to Lassa, Ebola, Marburg, Crimean-Congo fever viruses			devices and safe work practices, 2) hand hygiene; 3) barrier protection against blood and body fluids upon entry into room (single gloves and fluid-resistant or impermeable gown, face/eye protection with masks, goggles or face shields); and 4) appropriate waste handling. Use N95 or higher respirators when performing aerosol-generating procedures. Largest viral load in final stages of illness when hemorrhage may occur; additional PPE, including double gloves, leg and shoe coverings may be used, especially in resource-limited settings where options for cleaning and laundry are limited. Notify public health officials immediately if Ebola is suspected [212, 314, 740, 772] Also see Table 3 for Ebola as a bioterrorism agent
Viral respiratory diseases (not covered elsewhere)			
Adults	S		
Infants and young children (see respiratory infectious disease, acute)			
Whooping cough (see pertussis)			
Wound infections			
Major	C	DI	No dressing or dressing does not contain drainage adequately
Minor or limited	S		Dressing covers and contains drainage adequately
Yersinia enterocolitica gastroenteritis (see gastroenteritis)			
Zoster (varicella-zoster) (see herpes zoster)			
Zygomycosis (phycomycosis, mucormycosis)	S		Not transmitted person-to-person

TABLE 1. HISTORY OF GUIDELINES FOR ISOLATION PRECAUTIONS IN HOSPITALS*

YEAR (Ref)	DOCUMENT ISSUED	COMMENT
1970 [1099]	Isolation Techniques for Use in Hospitals, 1st ed.	- Introduced seven isolation precaution categories with color-coded cards: Strict, Respiratory, Protective, Enteric, Wound and Skin, Discharge, and Blood - No user decision-making required - Simplicity a strength; over isolation prescribed for some infections
1975 [1100]	Isolation Techniques for Use in Hospitals, 2nd ed.	- Same conceptual framework as 1st edition
1983 [1101]	CDC Guideline for Isolation Precautions in Hospitals	- Provided two systems for isolation: category-specific and disease-specific - Protective Isolation eliminated; Blood Precautions expanded to include Body Fluids - Categories included Strict, Contact, Respiratory, AFB, Enteric, Drainage/Secretion, Blood and Body Fluids - Emphasized decision-making by users
1985-88 [780, 896]	Universal Precautions	- Developed in response to HIV/AIDS epidemic - Dictated application of Blood and Body Fluid precautions to all patients, regardless of infection status - Did not apply to feces, nasal secretions, sputum, sweat, tears, urine, or vomitus unless contaminated by visible blood - Added personal protective equipment to protect HCWs from mucous membrane exposures - Handwashing recommended immediately after glove removal - Added specific recommendations for handling needles and other sharp devices; concept became integral to OSHA's 1991 rule on occupational exposure to blood-borne pathogens in healthcare settings

1987 [1102]	Body Substance Isolation	- Emphasized avoiding contact with all moist and potentially infectious body substances except sweat even if blood not present - Shared some features with Universal Precautions - Weak on infections transmitted by large droplets or by contact with dry surfaces - Did not emphasize need for special ventilation to contain airborne infections - Handwashing after glove removal not specified in the absence of visible soiling
1996 [1]	Guideline for Isolation Precautions in Hospitals	- Prepared by the Healthcare Infection Control Practices Advisory Committee (HICPAC) - Melded major features of Universal Precautions and Body Substance Isolation into Standard Precautions to be used with all patients at all times - Included three transmission-based precaution categories: airborne, droplet, and contact - Listed clinical syndromes that should dictate use of empiric isolation until an etiological diagnosis is established

* Derived from Garner ICHE 1996

TABLE 2. CLINICAL SYNDROMES OR CONDITIONS WARRANTING EMPIRIC TRANSMISSION-BASED PRECAUTIONS IN ADDITION TO STANDARD PRECAUTIONS PENDING CONFIRMATION OF DIAGNOSIS*

Clinical Syndrome or Condition†	Potential Pathogens‡	Empiric Precautions (Always includes Standard Precautions)
DIARRHEA		
Acute diarrhea with a likely infectious cause in an incontinent or diapered patient	Enteric pathogens§	Contact Precautions (pediatrics and adult)
MENINGITIS	*Neisseria meningitidis*	Droplet Precautions for first 24 hrs of antimicrobial therapy; mask and face protection for intubation
	Enteroviruses	Contact Precautions for infants and children
	M. tuberculosis	Airborne Precautions if pulmonary infiltrate Airborne Precautions plus Contact Precautions if potentially infectious draining body fluid present
RASH OR EXANTHEMS, GENERALIZED, ETIOLOGY UNKNOWN		
Petechial/ecchymotic with fever (general)	*Neisseria meningitides*	Droplet Precautions for first 24 hrs of antimicrobial therapy
- If positive history of travel to an area with an ongoing outbreak of VHF in the 10 days before onset of fever	Ebola, Lassa, Marburg viruses	Droplet Precautions plus Contact Precautions, with face/eye protection, emphasizing safety sharps and barrier precautions when blood exposure likely. Use N95 or higher respiratory protection when aerosol-generating procedure performed

Vesicular	Varicella-zoster, *herpes simplex,* variola (smallpox), vaccinia viruses Vaccinia virus	Airborne plus Contact Precautions; Contact Precautions only if *herpes simplex,* localized zoster in an immunocompetent host or vaccinia viruses most likely
Maculopapular with cough, coryza and fever	Rubeola (measles) virus	Airborne Precautions

Clinical Syndrome or Condition†	Potential Pathogens‡	Empiric Precautions (Always includes Standard Precautions)
RESPIRATORY INFECTIONS		
Cough/fever/upper lobe pulmonary infiltrate in an HIV-negative patient or a patient at low risk for human immunodeficiency virus (HIV) infection	*M. tuberculosis,* Respiratory viruses, *S. pneumoniae, S. aureus* (MSSA or MRSA)	Airborne Precautions plus Contact precautions
Cough/fever/pulmonary infiltrate in any lung location in an HIV-infected patient or a patient at high risk for HIV infection	*M. tuberculosis,* Respiratory viruses, *S. pneumoniae, S. aureus* (MSSA or MRSA)	Airborne Precautions plus Contact Precautions Use eye/face protection if aerosol-generating procedure performed or contact with respiratory secretions anticipated. If tuberculosis is unlikely and there are no AIIRs and/or respirators available, use Droplet Precautions instead of Airborne Precautions Tuberculosis more likely in HIV-infected individual than in HIV negative individual
Cough/fever/pulmonary infiltrate in any lung location in a patient with a history of recent travel (10-21 days) to countries with active outbreaks of SARS, avian influenza	*M. tuberculosis,* severe acute respiratory syndrome virus (SARS-CoV), avian influenza	Airborne plus Contact Precautions plus eye protection. If SARS and tuberculosis unlikely, use Droplet Precautions instead of Airborne Precautions.
Respiratory infections, particularly bronchiolitis and pneumonia, in infants and young children	Respiratory syncytial virus, parainfluenza virus, adenovirus, influenza virus, Human metapneumovirus	Contact plus Droplet Precautions; Droplet Precautions may be discontinued when adenovirus and influenza have been ruled out

Skin or Wound Infection		
Abscess or draining wound that cannot be covered	*Staphylococcus aureus* (MSSA or MRSA), group A streptococcus	Contact Precautions Add Droplet Precautions for the first 24 hours of appropriate antimicrobial therapy if invasive Group A streptococcal disease is suspected

* Infection control professionals should modify or adapt this table according to local conditions. To ensure that appropriate empiric precautions are implemented always, hospitals must have systems in place to evaluate patients routinely according to these criteria as part of their preadmission and admission care.

† Patients with the syndromes or conditions listed below may present with atypical signs or symptoms (e.g.neonates and adults with pertussis may not have paroxysmal or severe cough). The clinician's index of suspicion should be guided by the prevalence of specific conditions in the community, as well as clinical judgment.

‡ The organisms listed under the column "Potential Pathogens" are not intended to represent the complete, or even most likely, diagnoses, but rather possible etiologic agents that require additional precautions beyond Standard Precautions until they can be ruled out.

§ These pathogens include enterohemorrhagic *Escherichia coli* O157:H7, *Shigella spp,* hepatitis A virus, noroviruses, rotavirus, *C. difficile*.

TABLE 3.
INFECTION CONTROL CONSIDERATIONS FOR HIGH-PRIORITY (CDC CATEGORY A) DISEASES THAT MAY RESULT FROM BIOTERRORIST ATTACKS OR ARE CONSIDERED TO BE BIOTERRORIST THREATS
(www.bt.cdc.gov) [a]

[a] Abbreviations used in this table: RT = respiratory tract; GIT = gastrointestinal tract; CXR = chest x-ray; CT = computerized axial tomography; CSF = cerebrospinal fluid; and LD_{50} – lethal dose for 50% of experimental animals; HCWs = healthcare worker; BSL = biosafety level; PAPR = powered air purifying respirator; PCR = polymerase chain reaction; IHC = immunohistochemistry

Disease	**Anthrax**
Site(s) of Infection; Transmission Mode Cutaneous and inhalation disease have occurred in past bioterrorist incidents	**Cutaneous** (contact with spores);**RT** (inhalation of spores);**GIT** (ingestion of spores - rare) **Comment:** Spores can be inhaled into the lower respiratory tract. The infectious dose of *B. anthracis* in humans by any route is not precisely known. In primates, the LD50 (i.e., the dose required to kill 50% of animals) for an aerosol challenge with *B. anthracis* is estimated to be 8,000–50,000 spores; the infectious dose may be as low as 1-3 spores
Incubation Period	**Cutaneous**: 1 to12 days; **RT**: Usually 1 to 7 days but up to 43 days reported; **GIT**: 15-72 hours
Clinical Features	**Cutaneous:** Painless, reddish papule, which develops a central vesicle or bulla in 1-2 days; over next 3-7 days lesion becomes pustular, and then necrotic, with black eschar; extensive surrounding edema. **RT:** initial flu-like illness for 1-3 days with headache, fever, malaise, cough; by day 4 severe dyspnea and shock, and is usually fatal (85%-90% if untreated; meningitis in 50% of RT cases. **GIT**: ; if intestinal form, necrotic, ulcerated edematous lesions develop in intestines with fever, nausea and vomiting, progression to hematemesis and bloody diarrhea; 25-60% fatal
Diagnosis	**Cutaneous:** Swabs of lesion (under eschar) for IHC, PCR and culture; punch biopsy for IHC, PCR and culture; vesicular fluid aspirate for Gram stain and culture; blood culture if systemic symptoms; acute and

	convalescent sera for ELISA serology **RT:** CXR or CT demonstrating wide mediastinal widening and/or pleural effusion, hilar abnormalities; blood for culture and PCR; pleural effusion for culture, PCR and IHC; CSF if meningeal signs present for IHC, PCR and culture; acute and convalescent sera for ELISA serology; pleural and/or bronchial biopsies IHC. **GIT:** blood and ascites fluid, stool samples, rectal swabs, and swabs of oropharyngeal lesions if present for culture, PCR and IHC
Infectivity	**Cutaneous:** Person-to-person transmission from contact with lesion of untreated patient possible, but extremely rare. **RT and GIT:** Person-to-person transmission does not occur. **Aerosolized powder, environmental exposures:** Highly infectious if aerosolized
Recommended Precautions	**Cutaneous**: Standard Precautions; Contact Precautions if uncontained copious drainage. **RT and GIT:** Standard Precautions. **Aerosolized powder, environmental exposures:** Respirator (N95 mask or PAPRs), protective clothing; decontamination of persons with powder on them (http://www.cdc.gov/mmwr/preview/mmwrhtml/mm5135a3.htm) **Hand hygiene:** Handwashing for 30-60 seconds with soap and water or 2% chlorhexidene gluconate after spore contact (alcohol handrubs inactive against spores [Weber DJ JAMA 2003; 289:1274]). **Post-exposure prophylaxis following environmental exposure**: 60 days of antimicrobials (either doxycycline, ciprofloxacin, or levofloxacin) and post-exposure vaccine under IND

Disease	**Botulism**
Site(s) of Infection; Transmission Mode	**GIT:** Ingestion of toxin-containing food, **RT:** Inhalation of toxin containing aerosol cause disease. **Comment:** Toxin ingested or potentially delivered by aerosol in bioterrorist incidents. LD_{50} for type A is 0.001 μg/ml/kg.
Incubation Period	1-5 days.
Clinical Features	Ptosis, generalized weakness, dizziness, dry mouth and throat, blurred vision, diplopia, dysarthria, dysphonia, and dysphagia followed by symmetrical descending paralysis and respiratory failure.

Diagnosis	Clinical diagnosis; identification of toxin in stool, serology unless toxin-containing material available for toxin neutralization bioassays.
Infectivity	Not transmitted from person to person. Exposure to toxin necessary for disease.
Recommended Precautions	Standard Precautions.
Disease	**Ebola Hemorrhagic Fever**
Site(s) of Infection; Transmission Mode	As a rule infection develops after exposure of mucous membranes or RT, or through broken skin or percutaneous injury.
Incubation Period	2-19 days, usually 5-10 days
Clinical Features	Febrile illnesses with malaise, myalgias, headache, vomiting and diarrhea that are rapidly complicated by hypotension, shock, and hemorrhagic features. Massive hemorrhage in < 50% pts.
Diagnosis	Etiologic diagnosis can be made using RT-PCR, serologic detection of antibody and antigen, pathologic assessment with immunohistochemistry and viral culture with EM confirmation of morphology,
Infectivity	Person-to-person transmission primarily occurs through unprotected contact with blood and body fluids; percutaneous injuries (e.g., needlestick) associated with a high rate of transmission; transmission in healthcare settings has been reported but is prevented by use of barrier precautions.
Recommended Precautions	**Hemorrhagic fever specific barrier precautions**: If disease is believed to be related to intentional release of a bioweapon, epidemiology of transmission is unpredictable pending observation of disease transmission. Until the nature of the pathogen is understood and its transmission pattern confirmed, Standard, Contact and Airborne Precautions should be used. Once the pathogen is characterized, if the epidemiology of transmission is consistent with natural disease, Droplet Precautions can be substituted for Airborne Precautions. Emphasize: 1) use of sharps safety devices and safe work practices, 2) hand hygiene; 3) barrier protection against blood and body fluids upon entry into room (single gloves and fluid-resistant or impermeable gown, face/eye protection with masks, goggles or face shields); and 4) appropriate waste handling. Use N95 or higher respirators when performing aerosol-generating procedures. In settings where AIIRs are unavailable or the large numbers of patients cannot be accommodated by existing AIIRs, observe Droplet Precautions (plus Standard Precautions and Contact Precautions) and segregate patients from those not suspected of VHF infection. Limit blooddraws to those essential to care. See text for discussion and Appendix A for recommendations for naturally

	occurring VHFs.

Disease	Plague[2]
Site(s) of Infection; Transmission Mode	**RT:** Inhalation of respiratory droplets. **Comment**: Pneumonic plague most likely to occur if used as a biological weapon, but some cases of bubonic and primary septicemia may also occur. Infective dose 100 to 500 bacteria
Incubation Period	1 to 6, usually 2 to 3 days.
Clinical Features	Pneumonic: fever, chills, headache, cough, dyspnea, rapid progression of weakness, and in a later stage hemoptysis, circulatory collapse, and bleeding diathesis
Diagnosis	Presumptive diagnosis from Gram stain or Wayson stain of sputum, blood, or lymph node aspirate; definitive diagnosis from cultures of same material, or paired acute/convalescent serology.
Infectivity	Person-to-person transmission occurs via respiratory droplets risk of transmission is low during first 20-24 hours of illness and requires close contact. Respiratory secretions probably are not infectious within a few hours after initiation of appropriate therapy.
Recommended Precautions	Standard Precautions, Droplet Precautions until patients have received 48 hours of appropriate therapy. **Chemoprophylaxis:** Consider antibiotic prophylaxis for HCWs with close contact exposure.

[2] Pneumonic plague is not as contagious as is often thought. Historical accounts and contemporary evidence indicate that persons with plague usually only transmit the infection when the disease is in the end stage. These persons cough copious amounts of bloody sputum that contains many plague bacteria. Patients in the early stage of primary pneumonic plague (approximately the first 20–24 h) apparently pose little risk [1, 2]. Antibiotic medication rapidly clears the sputum of plague bacilli, so that a patient generally is not infective within hours after initiation of effective antibiotic treatment [3]. This means that in modern times many patients will never reach a stage where they pose a significant risk to others. Even in the end stage of disease, transmission only occurs after close contact. Simple protective measures, such as wearing masks, good hygiene, and avoiding close contact, have been effective to interrupt transmission during many pneumonic plague outbreaks [2]. In the United States, the last known cases of person to person transmission of pneumonic plague occurred in 1925 [2].

1. Wu L-T. A treatise on pneumonic plague. Geneva: League of Nations, 1926. III. Health.

2. Kool JL. Risk of person to person transmission of pneumonic plague. Clinical Infectious Diseases, 2005; 40 (8): 1166-1172

3. Butler TC. Plague and other Yersinia infections. In: Greenough WB, ed. Current topics in infectious disease. New York: Plenum Medical Book Company, 1983.

Disease	Smallpox
Site(s) of Infection; Transmission Mode	**RT** Inhalation of droplet or, rarely, aerosols; and skin lesions (contact with virus). **Comment:** If used as a biological weapon, natural disease, which has not occurred since 1977, will likely result.
Incubation Period	7 to 19 days (mean 12 days)
Clinical Features	Fever, malaise, backache, headache, and often vomiting for 2-3 days; then generalized papular or maculopapular rash (more on face and extremities), which becomes vesicular (on day 4 or 5) and then pustular; lesions all in same stage.
Diagnosis	Electron microscopy of vesicular fluid or culture of vesicular fluid by WHO approved laboratory (CDC); detection by PCR available only in select LRN labs, CDC and USAMRID
Infectivity	Secondary attack rates up to 50% in unvaccinated persons; infected persons may transmit disease from time rash appears until all lesions have crusted over (about 3 weeks); greatest infectivity during first 10 days of rash.
Recommended Precautions	Combined use of Standard, Contact, and Airborne Precautions[b] until all scabs have separated (3-4 weeks). Only immune HCWs to care for pts; post-exposure vaccine within 4 days. **Vaccinia:** HCWs cover vaccination site with gauze and semi-permeable dressing until scab separates (≥21 days). Observe hand hygiene. **Adverse events with virus-containing lesions:** Standard plus Contact Precautions until all lesions crusted

[b] Transmission by the airborne route is a rare event; Airborne Precautions is recommended when possible, but in the event of mass exposures, barrier precautions and containment within a designated area are most important [204, 212].

[c] Vaccinia adverse events with lesions containing infectious virus include inadvertent autoinoculation, ocular lesions (blepharitis, conjunctivitis), generalized vaccinia, progressive vaccinia, eczema vaccinatum; bacterial superinfection also requires addition of contact precautions if exudates cannot be contained [216, 217].

Disease	Tularemia
Site(s) of Infection; Transmission Mode	**RT:** Inhalation of aerosolized bacteria. **GIT:** Ingestion of food or drink contaminated with aerosolized bacteria. **Comment:** Pneumonic or typhoidal disease likely to occur after bioterrorist event using aerosol delivery. Infective dose 10-50 bacteria
Incubation Period	2 to 10 days, usually 3 to 5 days
Clinical Features	Pneumonic: malaise, cough, sputum production, dyspnea; Typhoidal: fever, prostration, weight loss and frequently an associated pneumonia.
Diagnosis	Diagnosis usually made with serology on acute and convalescent serum specimens; bacterium can be detected by PCR (LRN) or isolated from blood and other body fluids on cysteine-enriched media or mouse inoculation.
Infectivity	Person-to-person spread is rare. Laboratory workers who encounter/handle cultures of this organism are at high risk for disease if exposed.
Recommended Precautions	Standard Precautions

TABLE 4.
RECOMMENDATIONS FOR APPLICATION OF STANDARD PRECAUTIONS FOR THE CARE OF ALL PATIENTS IN ALL HEALTHCARE SETTINGS
(See Sections II.D.-II.J. and III.A.1)

COMPONENT	RECOMMENDATIONS
Hand hygiene	After touching blood, body fluids, secretions, excretions, contaminated items; immediately after removing gloves; between patient contacts.
Personal protective equipment (PPE)	
Gloves	For touching blood, body fluids, secretions, excretions, contaminated items; for touching mucous membranes and nonintact skin
Gown	During procedures and patient-care activities when contact of clothing/exposed skin with blood/body fluids, secretions, and excretions is anticipated..
Mask, eye protection (goggles), face shield*	During procedures and patient-care activities likely to generate splashes or sprays of blood, body fluids, secretions, especially suctioning, endotracheal intubation
Soiled patient-care equipment	Handle in a manner that prevents transfer of microorganisms to others and to the environment; wear gloves if visibly contaminated; perform hand hygiene.
Environmental control	Develop procedures for routine care, cleaning, and disinfection of environmental surfaces, especially frequently touched surfaces in patient-care areas.
Textiles and laundry	Handle in a manner that prevents transfer of microorganisms to others and to the environment
Needles and other sharps	Do not recap, bend, break, or hand-manipulate used needles; if recapping is required, use a one-handed scoop technique only; use safety features when available; place used sharps in puncture-resistant container
Patient resuscitation	Use mouthpiece, resuscitation bag, other ventilation devices to prevent contact with mouth and oral secretions

Patient placement	Prioritize for single-patient room if patient is at increased risk of transmission, is likely to contaminate the environment, does not maintain appropriate hygiene, or is at increased risk of acquiring infection or developing adverse outcome following infection.
Respiratory hygiene/cough etiquette (source containment of infectious respiratory secretions in symptomatic patients, beginning at initial point of encounter e.g., triage and reception areas in emergency departments and physician offices)	Instruct symptomatic persons to cover mouth/nose when sneezing/coughing; use tissues and dispose in no-touch receptacle; observe hand hygiene after soiling of hands with respiratory secretions; wear surgical mask if tolerated or maintain spatial separation, >3 feet if possible.

* * During aerosol-generating procedures on patients with suspected or proven infections transmitted by respiratory aerosols (e.g., SARS), wear a fit-tested N95 or higher respirator in addition to gloves, gown,and face/eye protection.

TABLE 5. COMPONENTS OF A PROTECTIVE ENVIRONMENT

(Adapted from MMWR 2003; 52 [RR-10])

I. Patients: allogeneic hematopoeitic stem cell transplant (HSCT) only

- Maintain in PE room except for required diagnostic or therapeutic procedures that cannot be performed in the room, e.g. radiology, operating room
- Respiratory protection e.g., N95 respirator, for the patient when leaving PE during periods of construction

II. Standard and Expanded Precautions

- Hand hygiene observed before and after patient contact
- Gown, gloves, mask NOT required for HCWs or visitors for routine entry into the room
- Use of gown, gloves, mask by HCWs and visitors according to Standard Precautions and as indicated for suspected or proven infections for which Transmission-Based Precautions are recommended

III. Engineering

- Central or point-of-use HEPA (99.97% efficiency) filters capable of removing particles 0.3 μm in diameter for supply (incoming) air
- Well-sealed rooms
  - Proper construction of windows, doors, and intake and exhaust ports
  - Ceilings: smooth, free of fissures, open joints, crevices
  - Walls sealed above and below the ceiling
  - If leakage detected, locate source and make necessary repairs
- Ventilation to maintain ≥12 ACH
- Directed air flow: air supply and exhaust grills located so that clean, filtered air enters from one side of the room, flows across the patient's bed, exits on opposite side of the room
- Positive room air pressure in relation to the corridor
  - Pressure differential of >2.5 Pa [0.01" water gauge]
- Monitor and document results of air flow patterns daily using visual methods (e.g., flutter strips, smoke tubes) or a hand held pressure gauge
- Self-closing door on all room exits
- Maintain back-up ventilation equipment (e.g., portable units for fans or filters) for emergency provision of ventilation requirements for PE areas and take immediate steps to restore the fixed ventilation system
- For patients who require both a PE and Airborne Infection Isolation, use an anteroom to ensure proper air balance relationships and provide independent exhaust of contaminated air to the outside or place a HEPA filter in the exhaust duct. If an anteroom is not available, place patient in an AIIR and use portable ventilation units, industrial-grade HEPA filters to enhance filtration of spores.

IV. Surfaces

- Daily wet-dusting of horizontal surfaces using cloths moistened with EPA-registered hospital disinfectant/detergent
- Avoid dusting methods that disperse dust
- No carpeting in patient rooms or hallways
- No upholstered furniture and furnishings

V. Other

- No flowers (fresh or dried) or potted plants in PE rooms or areas
- Use vacuum cleaner equipped with HEPA filters when vacuum cleaning is necessary

Figure.
Example of Safe Donning and Removal of Personal Protective Equipment (PPE)

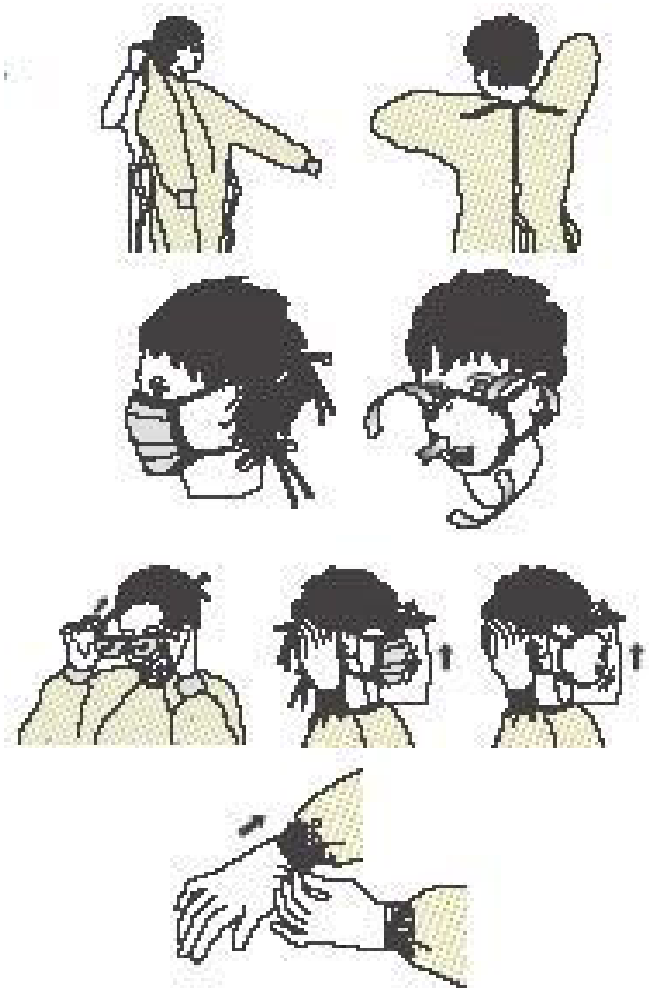
DONNING PPE

GOWN
- Fully cover torso from neck to knees, arms to end of wrist, and wrap around the back
- Fasten in back at neck and waist

MASK OR RESPIRATOR
- Secure ties or elastic band at middle of head and neck
- Fit flexible band to nose bridge
- Fit snug to face and below chin
- Fit-check respirator

GOGGLES/FACE SHIELD
- Put on face and adjust to fit

GLOVES
- Use non-sterile for isolation
- Select according to hand size
- Extend to cover wrist of isolation gown



SAFE WORK PRACTICES

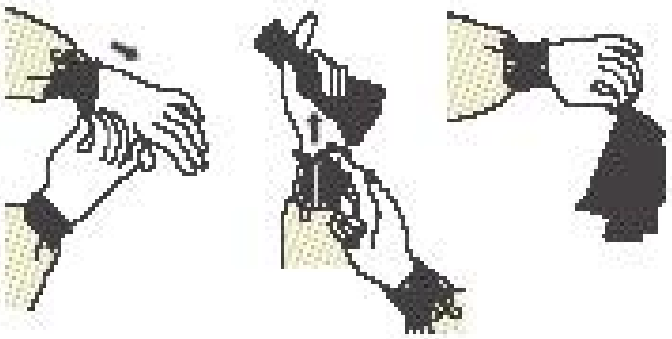
- Keep hands away from face
- Work from clean to dirty
- Limit surfaces touched
- Change when torn or heavily contaminated
- Perform hand hygiene

REMOVING PPE

Remove PPE at doorway before leaving patient room or in anteroom

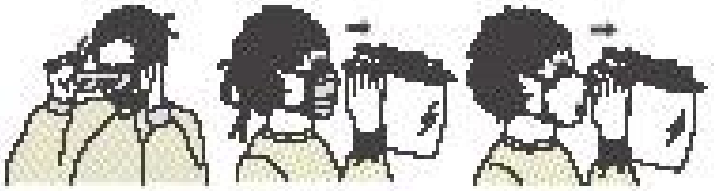
GLOVES

- Outside of gloves are contaminated!
- Grasp outside of glove with opposite gloved hand; peel off
- Hold removed glove in gloved hand
- Slide fingers of ungloved hand under remaining glove at wrist



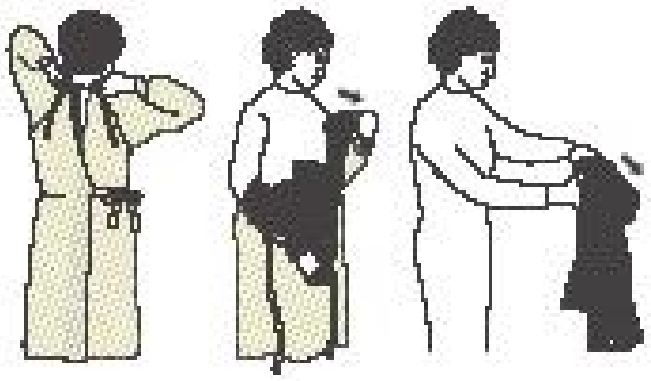
GOGGLES/FACE SHIELD

- Outside of goggles or face shield are contaminated!
- To remove, handle by "clean" head band or ear pieces
- Place in designated receptacle for reprocessing or in waste container



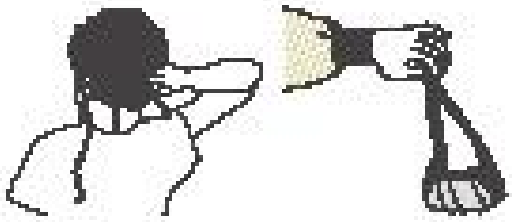
GOWN

- Gown front and sleeves are contaminated!
- Unfasten neck, then waist ties
- Remove gown using a peeling motion; pull gown from each shoulder toward the same hand
- Gown will turn inside out
- Hold removed gown away from body, roll into a bundle and discard into waste or linen receptacle



MASK OR RESPIRATOR

- Front of mask/respirator is contaminated – DO NOT TOUCH!
- Grasp ONLY bottom then top ties/elastics and remove
- Discard in waste container



HAND HYGIENE

Perform hand hygiene immediately after removing all PPE!

GLOSSARY

Airborne infection isolation room (AIIR). Formerly, negative pressure isolation room, an AIIR is a single-occupancy patient-care room used to isolate persons with a suspected or confirmed airborne infectious disease. Environmental factors are controlled in AIIRs to minimize the transmission of infectious agents that are usually transmitted from person to person by droplet nuclei associated with coughing or aerosolization of contaminated fluids. AIIRs should provide negative pressure in the room (so that air flows under the door gap into the room); **and** an air flow rate of 6-12 ACH (6 ACH for existing structures, 12 ACH for new construction or renovation); **and** direct exhaust of air from the room to the outside of the building or recirculation of air through a HEPA filter before retruning to circulation (MMWR 2005; 54 [RR-17]).

American Institute of Architects (AIA). A professional organization that develops standards for building ventilation, The "2001Guidelines for Design and Construction of Hospital and Health Care Facilities", the development of which was supported by the AIA, Academy of Architecture for Health, Facilities Guideline Institute, with assistance from the U.S. Department of Health and Human Services and the National Institutes of Health, is the primary source of guidance for creating airborne infection isolation rooms (AIIRs) and protective environments (www.aia.org/aah).

Ambulatory care settings. Facilities that provide health care to patients who do not remain overnight (e.g., hospital-based outpatient clinics, nonhospital-based clinics and physician offices, urgent care centers, surgicenters, free-standing dialysis centers, public health clinics, imaging centers, ambulatory behavioral health and substance abuse clinics, physical therapy and rehabilitation centers, and dental practices.

Bioaerosols. An airborne dispersion of particles containing whole or parts of biological entities, such as bacteria, viruses, dust mites, fungal hyphae, or fungal spores. Such aerosols usually consist of a mixture of mono-dispersed and aggregate cells, spores or viruses, carried by other materials, such as respiratory secretions and/or inert particles. Infectious bioaerosols (i.e., those that contain biological agents capable of causing an infectious disease) can be generated from human sources (e.g., expulsion from the respiratory tract during coughing, sneezing, talking or singing; during suctioning or wound irrigation), wet environmental sources (e.g. HVAC and cooling tower water with Legionella) or dry sources (e.g.,constuction dust with spores produced by *Aspergillus* spp.). Bioaerosols include large respiratory droplets and small droplet nuclei (Cole EC. AJIC 1998;26: 453-64).

Caregivers.. All persons who are not employees of an organization, are not paid, and provide or assist in providing healthcare to a patient (e.g., family member, friend) and acquire technical training as needed based on the tasks that must be performed.

Cohorting. In the context of this guideline, this term applies to the practice of grouping patients infected or colonized with the same infectious agent together to confine their care to one area and prevent contact with susceptible patients (cohorting patients). During outbreaks, healthcare personnel may be assigned to a cohort of patients to further limit opportunities for transmission (cohorting staff).

Colonization. Proliferation of microorganisms on or within body sites without detectable host immune response, cellular damage, or clinical expression. The presence of a microorganism within a host may occur with varying duration, but may become a source of potential transmission. In many instances, colonization and carriage are synonymous.

Droplet nuclei. Microscopic particles < 5 μm in size that are the residue of evaporated droplets and are produced when a person coughs, sneezes, shouts, or sings. These particles can remain suspended in the air for prolonged periods of time and can be carried on normal air currents in a room or beyond, to adjacent spaces or areas receiving exhaust air.

Engineering controls. Removal or isolation of a workplace hazard through technology. AIIRs, a Protective Environment, engineered sharps injury prevention devices and sharps containers are examples of engineering controls.

Epidemiologically important pathogens . Infectious agents that have one or more of the following characteristics: 1) are readily transmissible; 2) have a proclivity toward causing outbreaks; 3) may be associated with a severe outcome; or 4) are difficult to treat. Examples include *Acinetobacter sp., Aspergillus* sp.*, Burkholderia cepacia*, *Clostridium difficile*, *Klebsiella* or *Enterobacter* sp., extended-spectrum-beta-lactamase producing gram negative bacilli [ESBLs], methicillin-resistant *Staphylococcus aureus* [MRSA]*, Pseudomonas aeruginosa,* vancomycin-resistant enterococci [VRE], methicillin resistant *Staphylococcus aureus* [MRSA]*,* vancomycin resistant *Staphylococcus aureus* [VRSA] influenza virus, respiratory syncytial virus [RSV], rotavirus, SARS-CoV, noroviruses and the hemorrhagic fever viruses).

Hand hygiene. A general term that applies to any one of the following: 1) handwashing with plain (nonantimicrobial) soap and water); 2) antiseptic handwash (soap containing antiseptic agents and water); 3) antiseptic handrub (waterless antiseptic product, most often alcohol-based, rubbed on all surfaces of hands); or 4) surgical hand antisepsis (antiseptic handwash or antiseptic handrub performed preoperatively by surgical personnel to eliminate transient hand flora and reduce resident hand flora) [559].

Healthcare-associated infection (HAI). An infection that develops in a patient who is cared for in any setting where healthcare is delivered (e.g., acute care hospital, chronic care facility, ambulatory clinic, dialysis center, surgicenter, home) and is related to receiving health care (i.e., was not incubating or present at the time healthcare was provided). In ambulatory and home settings, HAI would apply to any infection that is associated with a medical or surgical intervention. Since the geographic location of infection acquisition is often uncertain, the preferred term is considered to be healthcare-*associated* rather than healthcare-*acquired*.

Healthcare epidemiologist. A person whose primary training is medical (M.D., D.O.) and/or masters or doctorate-level epidemiology who has received advanced training in healthcare epidemiology. Typically these professionals direct or provide consultation to an infection control program in a hospital, long term care facility (LTCF), or healthcare delivery system (also see infection control professional).

Healthcare personnel, healthcare worker (HCW). All paid and unpaid persons who work in a healthcare setting (e.g. any person who has professional or technical training in a healthcare-related field and provides patient care in a healthcare setting or any person who provides services that support the delivery of healthcare such as dietary, housekeeping, engineering, maintenance personnel).

Hematopoietic stem cell transplantation (HSCT). Any transplantation of blood- or bone marrow-derived hematopoietic stem cells, regardless of donor type (e.g., allogeneic or autologous) or cell source (e.g., bone marrow, peripheral blood, or placental/umbilical cord blood); associated with periods of severe immunosuppression that vary with the source of the cells, the intensity of chemotherapy required, and the presence of graft versus host disease (MMWR 2000; 49: RR-10).

High-efficiency particulate air (HEPA) filter. An air filter that removes >99.97% of particles $\geq$ 0.3µm (the most penetrating particle size) at a specified flow rate of air. HEPA filters may be integrated into the central air handling systems, installed at the point of use above the ceiling of a room, or used as portable units (MMWR 2003; 52: RR-10).

Home care. A wide-range of medical, nursing, rehabilitation, hospice and social services delivered to patients in their place of residence (e.g., private residence, senior living center, assisted living facility). Home health-care services include care provided by home health aides and skilled nurses, respiratory therapists, dieticians, physicians, chaplains, and volunteers; provision of durable medical equipment; home infusion therapy; and physical, speech, and occupational therapy.

Immunocompromised patients. Those patients whose immune mechanisms are deficient because of congenital or acquired immunologic disorders (e.g., human immunodeficiency virus [HIV] infection, congenital immune deficiency syndromes), chronic diseases such as diabetes mellitus, cancer, emphysema, or cardiac failure, ICU care, malnutrition, and immunosuppressive therapy of another disease process [e.g., radiation, cytotoxic chemotherapy, anti-graft-rejection medication, corticosteroids, monoclonal antibodies directed against a specific component of the immune system]). The type of infections for which an immunocompromised patient has increased susceptibility is determined by the severity of immunosuppression and the specific component(s) of the immune system that is affected. Patients undergoing allogeneic HSCT and those with chronic graft versus host disease are considered the most vulnerable to HAIs. Immunocompromised states also make it more difficult to diagnose certain infections (e.g., tuberculosis) and are associated with more severe clinical disease states than persons with the same infection and a normal immune system.

Infection. The transmission of microorganisms into a host after evading or overcoming defense mechanisms, resulting in the organism's proliferation and invasion within host tissue(s). Host responses to infection may include clinical symptoms or may be subclinical, with manifestations of disease mediated by direct organisms pathogenesis and/or a function of cell-mediated or antibody responses that result in the destruction of host tissues.

Infection control and prevention professional (ICP). A person whose primary training is in either nursing, medical technology, microbiology, or epidemiology and who has acquired special training in infection control. Responsibilities may include collection, analysis, and feedback of infection data and trends to healthcare providers; consultation on infection risk assessment, prevention and control strategies; performance of education and training activities; implementation of evidence-based infection control practices or those mandated by regulatory and licensing agencies; application of epidemiologic principles to improve patient outcomes; participation in planning renovation and construction projects (e.g., to ensure appropriate containment of construction dust); evaluation of new products or procedures on patient outcomes; oversight of employee health services related to infection prevention; implementation of preparedness plans; communication within the healthcare setting, with local and state health departments, and with the community at large concerning infection control issues; and participation in research. Certification in infection control (CIC) is available through the Certification Board of Infection Control and Epidemiology.

***Infection control and prevention program*.** A multidisciplinary program that includes a group of activities to ensure that recommended practices for the prevention of healthcare-associated infections are implemented and followed by HCWs, making the healthcare setting safe from infection for patients and

healthcare personnel. The Joint Commission on Accreditation of Healthcare Organizations (JCAHO) requires the following five components of an infection control program for accreditation: 1) *surveillance*: monitoring patients and healthcare personnel for acquisition of infection and/or colonization; 2) *investigation:* identification and analysis of infection problems or undesirable trends; 3) *prevention*: implementation of measures to prevent transmission of infectious agents and to reduce risks for device- and procedure-related infections; 4) *control*: evaluation and management of outbreaks; and 5) *reporting:* provision of information to external agencies as required by state and federal law and regulation (www.jcaho.org). The infection control program staff has the ultimate authority to determine infection control policies for a healthcare organization with the approval of the organization's governing body.

Long-term care facilities (LTCFs). An array of residential and outpatient facilities designed to meet the bio-psychosocial needs of persons with sustained self-care deficits. These include skilled nursing facilities, chronic disease hospitals, nursing homes, foster and group homes, institutions for the developmentally disabled, residential care facilities, assisted living facilities, retirement homes, adult day health care facilities, rehabilitation centers, and long-term psychiatric hospitals.

Mask. A term that applies collectively to items used to cover the nose and mouth and includes both procedure masks and surgical masks (www.fda.gov/cdrh/ode/guidance/094.html#4).

Multidrug-resistant organisms (MDROs). In general, bacteria that are resistant to one or more classes of antimicrobial agents and usually are resistant to all but one or two commercially available antimicrobial agents (e.g., MRSA, VRE, extended spectrum beta-lactamase [ESBL]-producing or intrinsically resistant gram-negative bacilli) [176].

Nosocomial infection. A term that is derived from two Greek words "nosos" (disease) and "komeion" (to take care of) and refers to any infection that develops during or as a result of an admission to an acute care facility (hospital) and was not incubating at the time of admission.

Personal protective equipment (PPE). A variety of barriers used alone or in combination to protect mucous membranes, skin, and clothing from contact with infectious agents. PPE includes gloves, masks, respirators, goggles, face shields, and gowns.

***Procedure Mask*.** A covering for the nose and mouth that is intended for use in general patient care situations. These masks generally attach to the face with ear loops rather than ties or elastic. Unlike surgical masks, procedure masks are not regulated by the Food and Drug Administration.

Protective Environment. A specialized patient-care area, usually in a hospital, that has a positive air flow relative to the corridor (i.e., air flows from the room to the outside adjacent space). The combination of high-efficiency particulate air (HEPA) filtration, high numbers (≥12) of air changes per hour (ACH), and minimal leakage of air into the room creates an environment that can safely accommodate patients with a severely compromised immune system (e.g., those who have received allogeneic hemopoietic stem-cell transplant [HSCT]) and decrease the risk of exposure to spores produced by environmental fungi. Other components include use of scrubbable surfaces instead of materials such as upholstery or carpeting, cleaning to prevent dust accumulation, and prohibition of fresh flowers or potted plants.

Quasi-experimental studies. Studies to evaluate interventions but do not use randomization as part of the study design. These studies are also referred to as nonrandomized, pre-post-intervention study designs. These studies aim to demonstrate causality between an intervention and an outcome but cannot achieve the level of confidence concerning attributable benefit obtained through a randomized, controlled trial. In hospitals and public health settings, randomized control trials often cannot be implemented due to ethical, practical and urgency reasons; therefore, quasi-experimental design studies are used commonly. However, even if an intervention appears to be effective statistically, the question can be raised as to the possibility of alternative explanations for the result.. Such study design is used when it is not logistically feasible or ethically possible to conduct a randomized, controlled trial, (e.g., during outbreaks). Within the classification of quasi-experimental study designs, there is a hierarchy of design features that may contribute to validity of results (Harris et al. CID 2004:38: 1586).

Residential care setting. A facility in which people live, minimal medical care is delivered, and the psychosocial needs of the residents are provided for.

Respirator. A personal protective device worn by healthcare personnel to protect them from inhalation exposure to airborne infectious agents that are < 5 μm in size. These include infectious droplet nuclei from patients with *M. tuberculosis,* variola virus [smallpox], SARS-CoV), and dust particles that contain infectious particles, such as spores of environmental fungi (e.g., *Aspergillus* sp.). The CDC's National Institute for Occupational Safety and Health (NIOSH) certifies respirators used in healthcare settings (www.cdc.gov/niosh/topics/respirators/). The N95 disposable particulate, air purifying, respirator is the type used most commonly by healthcare personnel. Other respirators used include N-99 and N-100 particulate respirators, powered air-purifying respirators (PAPRS) with high efficiency filters; and non-powered full-facepiece elastomeric negative pressure respirators. A listing of NIOSH-approved respirators can be found at www.cdc.gov/niosh/npptl/respirators/disp_part/particlist.html. Respirators must be used in conjunction with a complete Respiratory Protection Program, as

required by the Occupational Safety and Health Administration (OSHA), that includes fit testing, training, proper selection of respirators, medical clearance and respirator maintenance.

Respiratory Hygiene/ Cough Etiquette. A combination of measures designed to minimize the transmission of respiratory pathogens via droplet or airborne routes in healthcare settings. The components of Respiratory Hygiene/Cough Etiquette are 1) covering the mouth and nose during coughing and sneezing, 2) using tissues to contain respiratory secretions with prompt disposal into a no-touch receptacle, 3) offering a surgical mask to persons who are coughing to decrease contamination of the surrounding environment, and 4) turning the head away from others and maintaining spatial separation, ideally >3 feet, when coughing. These measures are targeted to all patients with symptoms of respiratory infection and their accompanying family members or friends beginning at the point of initial encounter with a healthcare setting (e.g., reception/triage in emergency departments, ambulatory clinics, healthcare provider offices) [126] (Srinivasin A ICHE 2004; 25: 1020; www.cdc.gov/flu/professionals/infectioncontrol/resphygiene.htm).

Safety culture/climate. The shared perceptions of workers and management regarding the expectations of safety in the work environment. A hospital safety climate includes the following six organizational components: 1) senior management support for safety programs; 2) absence of workplace barriers to safe work practices; 3) cleanliness and orderliness of the worksite; 4) minimal conflict and good communication among staff members; 5) frequent safety-related feedback/training by supervisors; and 6) availability of PPE and engineering controls [620].

Source Control. The process of containing an infectious agent either at the portal of exit from the body or within a confined space. The term is applied most frequently to containment of infectious agents transmitted by the respiratory route but could apply to other routes of transmission, (e.g., a draining wound, vesicular or bullous skin lesions). Respiratory Hygiene/Cough Etiquette that encourages individuals to “cover your cough” and/or wear a mask is a source control measure. The use of enclosing devices for local exhaust ventilation (e.g., booths for sputum induction or administration of aerosolized medication) is another example of source control.

Standard Precautions. A group of infection prevention practices that apply to all patients, regardless of suspected or confirmed diagnosis or presumed infection status. Standard Precautions is a combination and expansion of Universal Precautions [780] and Body Substance Isolation [1102]. Standard Precautions is based on the principle that all blood, body fluids, secretions, excretions except sweat, nonintact skin, and mucous membranes may contain transmissible infectious agents. Standard Precautions includes hand hygiene, and depending on the anticipated exposure, use of gloves, gown, mask, eye protection, or face shield. Also, equipment or items in the patient environment

likely to have been contaminated with infectious fluids must be handled in a manner to prevent transmission of infectious agents, (e.g. wear gloves for handling, contain heavily soiled equipment, properly clean and disinfect or sterilize reusable equipment before use on another patient).

Surgical mask. A device worn over the mouth and nose by operating room personnel during surgical procedures to protect both surgical patients and operating room personnel from transfer of microorganisms and body fluids. Surgical masks also are used to protect healthcare personnel from contact with large infectious droplets (>5 μm in size). According to draft guidance issued by the Food and Drug Administration on May 15, 2003, surgical masks are evaluated using standardized testing procedures for fluid resistance, bacterial filtration efficiency, differential pressure (air exchange), and flammability in order to mitigate the risks to health associated with the use of surgical masks. These specifications apply to any masks that are labeled surgical, laser, isolation, or dental or medical procedure (www.fda.gov/cdrh/ode/guidance/094.html#4). Surgical masks do not protect against inhalation of small particles or droplet nuclei and should not be confused with particulate respirators that are recommended for protection against selected airborne infectious agents, (e.g., *Mycobacterium tuberculosis)*.

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GUIDELINE FOR PREVENTION OF CATHETER-ASSOCIATED URINARY TRACT INFECTIONS 2009

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Abbreviations

ADL	Activities of daily living
APACHE II	Acute Physiology and Chronic Health Evaluation II
ASA	American Society of Anesthesiologists
ASB	Asymptomatic bacteriuria
BUN	Blood urea nitrogen
CAUTI	Catheter-associated urinary tract infection
CDC	Centers for Disease Control and Prevention
CFU	Colony-forming units
CI	Confidence interval
CIC	Clean intermittent catheterization
CICU	Coronary intensive care unit
COPD	Chronic obstructive pulmonary disease
ED	Emergency department
F/U	Follow-up
GRADE	Grading of Recommendations Assessment, Development, and Evaluation system
Hb	Hemoglobin concentration
HICPAC	Healthcare Infection Control Practices Advisory Committee
H/O	History of
HPF	High power field
HR	Hazard ratio
ICU	Intensive care unit
IDR	Incidence-density ratio
LOS	Length of stay
MDR	Multi-drug resistant
MICU	Medical intensive care unit
NHSN	National Healthcare Safety Network
NIH	National Institutes of Health
NS	Not significant
OBS	Observational controlled study
OR	Odds ratio
P	P value
PACU	Post-anesthesia care unit
PVC	Polyvinyl chloride

RCT	Randomized controlled trial
RD	Risk difference
RH	Relative hazard
RR	Relative risk
SAPS II	Simplified Acute Physiology Score II
SICU	Surgical intensive care unit
SR	Systematic review
SUTI	Symptomatic urinary tract infection
TMP/SMX	Trimethoprim/sulfamethoxazole
TURP	Transurethral resection of prostate
UTI	Urinary tract infection
VAS	Visual analog scale
WMD	Weighted mean difference

I. Executive Summary

This guideline updates and expands the original Centers for Disease Control and Prevention (CDC) Guideline for Prevention of Catheter-associated Urinary Tract Infections (CAUTI) published in 1981. Several developments necessitated revision of the 1981 guideline, including new research and technological advancements for preventing CAUTI, increasing need to address patients in non-acute care settings and patients requiring long-term urinary catheterization, and greater emphasis on prevention initiatives as well as better defined goals and metrics for outcomes and process measures. In addition to updating the previous guideline, this revised guideline reviews the available evidence on CAUTI prevention for patients requiring chronic indwelling catheters and individuals who can be managed with alternative methods of urinary drainage (e.g., intermittent catheterization). The revised guideline also includes specific recommendations for implementation, performance measurement, and surveillance. Although the general principles of CAUTI prevention have not changed from the previous version, the revised guideline provides clarification and more specific guidance based on a defined, systematic review of the literature through July 2007. For areas where knowledge gaps exist, recommendations for further research are listed. Finally, the revised guideline outlines high-priority recommendations for CAUTI prevention in order to offer guidance for implementation.

This document is intended for use by infection prevention staff, healthcare epidemiologists, healthcare administrators, nurses, other healthcare providers, and persons responsible for developing, implementing, and evaluating infection prevention and control programs for healthcare settings across the continuum of care. The guideline can also be used as a resource for societies or organizations that wish to develop more detailed implementation guidance for prevention of CAUTI.

Our goal was to develop a guideline based on a targeted systematic review of the best available evidence, with explicit links between the evidence and recommendations. To accomplish this, we used an adapted GRADE system approach for evaluating quality of evidence and determining strength of recommendations. The methodology, structure, and components of this guideline are approved by HICPAC and will be used for subsequent guidelines issued by HICPAC. A more detailed description of our approach is available in the Methods section.

To evaluate the evidence on preventing CAUTI, we examined data addressing three key questions and related subquestions:

1. Who should receive urinary catheters?
    A. When is urinary catheterization necessary?
    B. What are the risk factors for CAUTI?
    C. What populations are at highest risk of mortality related to urinary catheters?
2. For those who may require urinary catheters, what are the best practices? Specifically, what are the risks and benefits associated with:
    A. Different approaches to catheterization?
    B. Different catheters or collecting systems?
    C. Different catheter management techniques?
    D. Different systems interventions (i.e., quality improvement programs)?
3. What are the best practices for preventing CAUTI associated with obstructed urinary catheters?

Evidence addressing the key questions was used to formulate recommendations, and explicit links between the evidence and recommendations are available in the Evidence Review in the body of the guideline and Evidence Tables and GRADE Tables in the Appendices. **It is important to note that Category I recommendations are all considered strong recommendations and should be equally implemented;** it is only the *quality* of the evidence underlying the recommendation that distinguishes between levels A and B. Category IC recommendations are required by state or federal regulation and may have any level of supporting evidence.

The categorization scheme used in this guideline is presented in Table 1 in the Summary of Recommendations and described further in the Methods section.

The Summary of Recommendations is organized as follows: 1) recommendations for who should receive indwelling urinary catheters (or, for certain populations, alternatives to indwelling catheters); 2) recommendations for catheter insertion; 3) recommendations for catheter maintenance; 4) quality improvement programs to achieve appropriate placement, care, and removal of catheters; 5) administrative infrastructure required; and 6) surveillance strategies.

The Implementation and Audit section includes a prioritization of recommendations (i.e., high-priority recommendations that are essential for every healthcare facility), organized by modules, in order to provide facilities more guidance on implementation of these guidelines. A list of recommended performance measures that can potentially be used for internal reporting purposes is also included.

Areas in need of further research identified during the evidence review are outlined in the Recommendations for Further Research. This section includes guidance for specific methodological approaches that should be used in future studies.

Readers who wish to examine the primary evidence underlying the recommendations are referred to the Evidence Review in the body of the guideline, and the Evidence Tables and GRADE Tables in the Appendices. The Evidence Review includes narrative summaries of the data presented in the Evidence Tables and GRADE Tables. The Evidence Tables include all study-level data used in the guideline, and the GRADE Tables assess the overall quality of evidence for each question. The Appendices also contain a clearly delineated search strategy that will be used for periodic updates to ensure that the guideline remains a timely resource as new information becomes available.

II. Summary of Recommendations

Table 1. Modified HICPAC Categorization Scheme* for Recommendations	
Category IA	A strong recommendation supported by high to moderate quality† evidence suggesting net clinical benefits or harms
Category IB	A strong recommendation supported by low quality evidence suggesting net clinical benefits or harms or an accepted practice (e.g., aseptic technique) supported by low to very low quality evidence
Category IC	A strong recommendation required by state or federal regulation.
Category II	A weak recommendation supported by any quality evidence suggesting a trade off between clinical benefits and harms
No recommendation/ unresolved issue	Unresolved issue for which there is low to very low quality evidence with uncertain trade offs between benefits and harms

* Please refer to Methods (p.32) for implications of Category designations
†Please refer to Methods (p. 29-30) for process used to grade quality of evidence

I. Appropriate Urinary Catheter Use

A. Insert catheters only for appropriate indications (see Table 2 for guidance), and leave in place only as long as needed. **(Category IB)** (Key Questions 1B and 2C)

1. Minimize urinary catheter use and duration of use in all patients, particularly those at higher risk for CAUTI or mortality from catheterization such as women, the elderly, and patients with impaired immunity.**(Category IB)** (Key Questions 1B and 1C)

2. Avoid use of urinary catheters in patients and nursing home residents for management of incontinence. **(Category IB)** (Key Question 1A)

    a. Further research is needed on periodic (e.g., nighttime) use of external catheters (e.g., condom catheters) in incontinent patients or residents and the use of catheters to prevent skin breakdown. **(No recommendation/unresolved issue)** (Key Question 1A)

3. Use urinary catheters in operative patients only as necessary, rather than routinely. **(Category IB)** (Key Question 1A)

4. For operative patients who have an indication for an indwelling catheter, remove the catheter as soon as possible postoperatively, preferably within 24 hours, unless there are appropriate indications for continued use. **(Category IB)** (Key Questions 2A and 2C)

Table 2. **A. Examples of Appropriate Indications for Indwelling Urethral Catheter Use** [1-4]
Patient has acute urinary retention or bladder outlet obstruction
Need for accurate measurements of urinary output in critically ill patients
Perioperative use for selected surgical procedures: • Patients undergoing urologic surgery or other surgery on contiguous structures of the genitourinary tract • Anticipated prolonged duration of surgery (catheters inserted for this reason should be removed in PACU) • Patients anticipated to receive large-volume infusions or diuretics during surgery • Need for intraoperative monitoring of urinary output
To assist in healing of open sacral or perineal wounds in incontinent patients
Patient requires prolonged immobilization (e.g., potentially unstable thoracic or lumbar spine, multiple traumatic injuries such as pelvic fractures)
To improve comfort for end of life care if needed
B. Examples of *Inappropriate* Uses of Indwelling Catheters
As a substitute for nursing care of the patient or resident with incontinence
As a means of obtaining urine for culture or other diagnostic tests when the patient can voluntarily void
For prolonged postoperative duration without appropriate indications (e.g., structural repair of urethra or contiguous structures, prolonged effect of epidural anaesthesia, etc.)

Note: These indications are based primarily on expert consensus.

B. Consider using alternatives to indwelling urethral catheterization in selected patients when appropriate.

1. Consider using external catheters as an alternative to indwelling urethral catheters in cooperative male patients without urinary retention or bladder outlet obstruction. **(Category II)** (Key Question 2A)

2. Consider alternatives to chronic indwelling catheters, such as intermittent catheterization, in spinal cord injury patients. **(Category II)** (Key Question 1A)

3. Intermittent catheterization is preferable to indwelling urethral or suprapubic catheters in patients with bladder emptying dysfunction. **(Category II)** (Key Question 2A)

4. Consider intermittent catheterization in children with myelomeningocele and neurogenic bladder to reduce the risk of urinary tract deterioration. **(Category II)** (Key Question 1A)

5. Further research is needed on the benefit of using a urethral stent as an alternative to an indwelling catheter in selected patients with bladder outlet obstruction. **(No recommendation/unresolved issue)** (Key Question 1A)

6. Further research is needed on the risks and benefits of suprapubic catheters as an alternative to indwelling urethral catheters in selected patients requiring short- or long-term catheterization, particularly with respect to complications related to catheter insertion or the catheter site. **(No recommendation/unresolved issue)** (Key Question 2A)

II. Proper Techniques for Urinary Catheter Insertion

A. Perform hand hygiene immediately before and after insertion or any manipulation of the catheter device or site. **(Category IB)** (Key Question 2D)

B. Ensure that only properly trained persons (e.g., hospital personnel, family members, or patients themselves) who know the correct technique of aseptic catheter insertion and maintenance are given this responsibility. **(Category IB)** (Key Question 1B)

C. In the acute care hospital setting, insert urinary catheters using aseptic technique and sterile equipment. **(Category IB)**

    1. Use sterile gloves, drape, sponges, an appropriate antiseptic or sterile solution for periurethral cleaning, and a single-use packet of lubricant jelly for insertion. **(Category IB)**

    2. Routine use of antiseptic lubricants is not necessary. **(Category II)** (Key Question 2C)

    3. Further research is needed on the use of antiseptic solutions vs. sterile water or saline for periurethral cleaning prior to catheter insertion. **(No recommendation/unresolved issue)** (Key Question 2C)

D. In the non-acute care setting, clean (i.e., non-sterile) technique for intermittent catheterization is an acceptable and more practical alternative to sterile technique for patients requiring chronic intermittent catheterization. **(Category IA)** (Key Question 2A)

    1. Further research is needed on optimal cleaning and storage methods for catheters used for clean intermittent catheterization. **(No recommendation/unresolved issue)** (Key Question 2C)

E. Properly secure indwelling catheters after insertion to prevent movement and urethral traction. **(Category IB)**

F. Unless otherwise clinically indicated, consider using the smallest bore catheter possible, consistent with good drainage, to minimize bladder neck and urethral trauma. **(Category II)**

G. If intermittent catheterization is used, perform it at regular intervals to prevent bladder overdistension. **(Category IB)** (Key Question 2A)

H. Consider using a portable ultrasound device to assess urine volume in patients undergoing intermittent catheterization to assess urine volume and reduce unnecessary catheter insertions. **(Category II)** (Key Question 2C)

    1. If ultrasound bladder scanners are used, ensure that indications for use are clearly stated, nursing staff are trained in their use, and equipment is adequately cleaned and disinfected in between patients. **(Category IB)**

III. Proper Techniques for Urinary Catheter Maintenance

A. Following aseptic insertion of the urinary catheter, maintain a closed drainage system **(Category IB)** (Key Question 1B and 2B)

   1. If breaks in aseptic technique, disconnection, or leakage occur, replace the catheter and collecting system using aseptic technique and sterile equipment. **(Category IB)**

   2. Consider using urinary catheter systems with preconnected, sealed catheter-tubing junctions. **(Category II)** (Key Question 2B)

B. Maintain unobstructed urine flow. **(Category IB)** (Key Questions 1B and 2D)

   1. Keep the catheter and collecting tube free from kinking. **(Category IB)**

   2. Keep the collecting bag below the level of the bladder at all times. Do not rest the bag on the floor. **(Category I**B**)**

   3. Empty the collecting bag regularly using a separate, clean collecting container for each patient; avoid splashing, and prevent contact of the drainage spigot with the nonsterile collecting container. **(Category IB)**

C. Use Standard Precautions, including the use of gloves and gown as appropriate, during any manipulation of the catheter or collecting system. **(Category IB)**

D. Complex urinary drainage systems (utilizing mechanisms for reducing bacterial entry such as antiseptic-release cartridges in the drain port) are not necessary for routine use. **(Category II)** (Key Question 2B)

E. Changing indwelling catheters or drainage bags at routine, fixed intervals is not recommended. Rather, it is suggested to change catheters and drainage bags based on clinical indications such as infection, obstruction, or when the closed system is compromised. **(Category II)** (Key Question 2C)

F. Unless clinical indications exist (e.g., in patients with bacteriuria upon catheter removal post urologic surgery), do not use systemic antimicrobials routinely to prevent CAUTI in patients requiring either short or long-term catheterization. **(Category IB)** (Key Question 2C)

   1. Further research is needed on the use of urinary antiseptics (e.g., methenamine) to prevent UTI in patients requiring short-term catheterization. **(No recommendation/unresolved issue)** (Key Question 2C)

G. Do not clean the periurethral area with antiseptics to prevent CAUTI while the catheter is in place. Routine hygiene (e.g., cleansing of the meatal surface during daily bathing or showering) is appropriate. **(Category IB)** (Key Question 2C)

H. Unless obstruction is anticipated (e.g., as might occur with bleeding after prostatic or bladder surgery) bladder irrigation is not recommended. **(Category II)** (Key Question 2C)

1. If obstruction is anticipated, closed continuous irrigation is suggested to prevent obstruction. **(Category II)**

I. Routine irrigation of the bladder with antimicrobials is not recommended. **(Category II)** (Key Question 2C)

J. Routine instillation of antiseptic or antimicrobial solutions into urinary drainage bags is not recommended. **(Category II)** (Key Question 2C)

K. Clamping indwelling catheters prior to removal is not necessary. **(Category II)** (Key Question 2C)

L. Further research is needed on the use of bacterial interference (i.e., bladder inoculation with a nonpathogenic bacterial strain) to prevent UTI in patients requiring chronic urinary catheterization. **(No recommendation/unresolved issue)** (Key Question 2C)

Catheter Materials

M. If the CAUTI rate is not decreasing after implementing a comprehensive strategy to reduce rates of CAUTI, consider using antimicrobial/antiseptic-impregnated catheters. The comprehensive strategy should include, at a minimum, the high priority recommendations for urinary catheter use, aseptic insertion, and maintenance (see Section III. Implementation and Audit). **(Category IB)** (Key Question 2B)

1. Further research is needed on the effect of antimicrobial/antiseptic-impregnated catheters in reducing the risk of symptomatic UTI, their inclusion among the primary interventions, and the patient populations most likely to benefit from these catheters. **(No recommendation/unresolved issue)** (Key Question 2B)

N. Hydrophilic catheters might be preferable to standard catheters for patients requiring intermittent catheterization. **(Category II)** (Key Question 2B)

O. Silicone might be preferable to other catheter materials to reduce the risk of encrustation in long-term catheterized patients who have frequent obstruction. **(Category II)** (Key Question 3)

P. Further research is needed to clarify the benefit of catheter valves in reducing the risk of CAUTI and other urinary complications. **(No recommendation/unresolved issue)** (Key Question 2B)

Management of Obstruction

Q. If obstruction occurs and it is likely that the catheter material is contributing to obstruction, change the catheter. **(Category IB)**

R. Further research is needed on the benefit of irrigating the catheter with acidifying solutions or use of oral urease inhibitors in long-term catheterized patients who have frequent catheter obstruction. **(No recommendation/unresolved issue)** (Key Question 3)

S. Further research is needed on the use of a portable ultrasound device to evaluate for obstruction in patients with indwelling catheters and low urine output. **(No recommendation/unresolved issue)** (Key Question 2C)

T. Further research is needed on the use of methenamine to prevent encrustation in patients requiring chronic indwelling catheters who are at high risk for obstruction. **(No recommendation/unresolved issue)** (Key Question 2C)

Specimen Collection

U. Obtain urine samples aseptically. **(Category IB)**

    1. If a small volume of fresh urine is needed for examination (i.e., urinalysis or culture), aspirate the urine from the needleless sampling port with a sterile syringe/cannula adapter after cleansing the port with a disinfectant. **(Category IB)**

    2. Obtain large volumes of urine for special analyses (not culture) aseptically from the drainage bag. **(Category IB)**

Spatial Separation of Catheterized Patients

V. Further research is needed on the benefit of spatial separation of patients with urinary catheters to prevent transmission of pathogens colonizing urinary drainage systems. **(No recommendation/unresolved issue)** (Key Question 2D)

IV. Quality Improvement Programs

A. Implement quality improvement (QI) programs or strategies to enhance appropriate use of indwelling catheters and to reduce the risk of CAUTI based on a facility risk assessment. **(Category IB)** (Key Question 2D)

    The purposes of QI programs should be: 1) to assure appropriate utilization of catheters 2) to identify and remove catheters that are no longer needed (e.g., daily review of their continued need) and 3) to ensure adherence to hand hygiene and proper care of catheters. Examples of programs that have been demonstrated to be effective include:

    1. A system of alerts or reminders to identify all patients with urinary catheters and assess the need for continued catheterization

    2. Guidelines and protocols for nurse-directed removal of unnecessary urinary catheters

    3. Education and performance feedback regarding appropriate use, hand hygiene, and catheter care

    4. Guidelines and algorithms for appropriate peri-operative catheter management, such as:

a. Procedure-specific guidelines for catheter placement and postoperative catheter removal

b. Protocols for management of postoperative urinary retention, such as nurse-directed use of intermittent catheterization and use of bladder ultrasound scanners

V. Administrative Infrastructure

A. Provision of guidelines

1. Provide and implement evidence-based guidelines that address catheter use, insertion, and maintenance. **(Category IB)**

a. Consider monitoring adherence to facility-based criteria for acceptable indications for indwelling urinary catheter use. **(Category II)**

B. Education and Training

1. Ensure that healthcare personnel and others who take care of catheters are given periodic in-service training regarding techniques and procedures for urinary catheter insertion, maintenance, and removal. Provide education about CAUTI, other complications of urinary catheterization, and alternatives to indwelling catheters. **(Category IB)**

2. When feasible, consider providing performance feedback to these personnel on what proportion of catheters they have placed meet facility-based criteria and other aspects related to catheter care and maintenance. **(Category II)**

C. Supplies

1. Ensure that supplies necessary for aseptic technique for catheter insertion are readily available. **(Category IB)**

D. System of documentation

1. Consider implementing a system for documenting the following in the patient record: indications for catheter insertion, date and time of catheter insertion, individual who inserted catheter, and date and time of catheter removal. **(Category II)**

a. Ensuring that documentation is accessible in the patient record and recorded in a standard format for data collection and quality improvement purposes is suggested. Electronic documentation that is searchable is preferable. **(Category II)**

E. Surveillance resources

1. If surveillance for CAUTI is performed, ensure that there are sufficient trained personnel and technology resources to support surveillance for urinary catheter use and outcomes. **(Category IB)**

VI. Surveillance

A. Consider surveillance for CAUTI when indicated by facility-based risk assessment. **(Category II)**

   1. Identify the patient groups or units on which to conduct surveillance based on frequency of catheter use and potential risk of CAUTI.

B. Use standardized methodology for performing CAUTI surveillance. **(Category IB)**

   1. Examples of metrics that should be used for CAUTI surveillance include:

      a. Number of CAUTI per 1000 catheter-days

      b. Number of bloodstream infections secondary to CAUTI per 1000 catheter-days

      c. Catheter utilization ratio: (urinary catheter days/patient days) x 100

   2. Use CDC/NHSN criteria for identifying patients who have symptomatic UTI (SUTI) (numerator data) (see NHSN Patient Safety Manual: http://www.cdc.gov/nhsn/library.html).

   3. For more information on metrics, please see the U.S. Department of Health & Human Services (HHS) Action Plan to Prevent Healthcare-Associated Infections: http://www.hhs.gov/ophs/initiatives/hai/infection.html.

C. Routine screening of catheterized patients for asymptomatic bacteriuria (ASB) is not recommended. **(Category II)** (Key Question 2D)

D. When performing surveillance for CAUTI, consider providing regular (e.g., quarterly) feedback of unit-specific CAUTI rates to nursing staff and other appropriate clinical care staff. **(Category II)** (Key Question 2D)

III. Implementation and Audit

Prioritization of Recommendations

In this section, the recommendations considered essential for *all* healthcare facilities caring for patients requiring urinary catheterization are organized into modules in order to provide more guidance to facilities on implementation of these guidelines. The high-priority recommendations were chosen by a consensus of experts based on strength of recommendation as well as on the likely impact of the strategy in preventing CAUTI. The administrative functions and infrastructure listed above in the summary of recommendations are necessary to accomplish the high priority recommendations and are therefore critical to the success of a prevention program. In addition, quality improvement programs should be implemented as an active approach to accomplishing these recommendations and when process and outcome measure goals are not being met based on internal reporting.

Priority Recommendations for Appropriate Urinary Catheter Use (Module 1)

- Insert catheters only for appropriate indications (see Table 2), and leave in place only as long as needed. **(Category IB)**
    - Avoid use of urinary catheters in patients and nursing home residents for management of incontinence. **(Category IB)**
    - For operative patients who have an indication for an indwelling catheter, remove the catheter as soon as possible postoperatively, preferably within 24 hours, unless there are appropriate indications for continued use. **(Category IB)**

Priority Recommendations for Aseptic Insertion of Urinary Catheters (Module 2)

- Ensure that only properly trained persons (e.g., hospital personnel, family members, or patients themselves) who know the correct technique of aseptic catheter insertion and maintenance are given this responsibility. **(Category IB)**
- In the acute care hospital setting, insert catheters using aseptic technique and sterile equipment. **(Category IB)**

Priority Recommendations for Proper Urinary Catheter Maintenance (Module 3)

- Following aseptic insertion of the urinary catheter, maintain a closed drainage system **(Category IB)**
- Maintain unobstructed urine flow. **(Category IB)**

Performance Measures

A. Internal Reporting. Consider reporting both process and outcome measures to senior administrative, medical, and nursing leadership and clinicians who care for patients at risk for CAUTI. **(Category II)**

1. Examples of process measures:
    a) Compliance with educational program: Calculate percent of personnel who have proper training:
        - Numerator: number of personnel who insert urinary catheters and who have proper training
        - Denominator: number of personnel who insert urinary catheters
        - Standardization factor: 100 (i.e., multiply by 100 so that measure is expressed as a percentage)

b) Compliance with documentation of catheter insertion and removal dates: Conduct random audits of selected units and calculate compliance rate:
    - Numerator: number of patients on unit with catheters with proper documentation of insertion and removal dates
    - Denominator: number of patients on the unit with a catheter in place at some point during admission
    - Standardization factor: 100 (i.e., multiply by 100 so that measure is expressed as a percentage)

c) Compliance with documentation of indication for catheter placement: Conduct random audits of selected units and calculate compliance rate
    - Numerator: number of patients on unit with catheters with proper documentation of indication
    - Denominator: number of patients on the unit with catheter in place
    - Standardization factor: 100 (i.e., multiply by 100 so that measure is expressed as a percentage)

2. Recommended outcome measures:

a) Rates of CAUTI: Use NHSN definitions (see http://www.cdc.gov/nhsn/library.html). Measurement of rates allows an individual facility to gauge the longitudinal impact of implementation of prevention strategies:
    - Numerator: number of CAUTIs in each location monitored
    - Denominator: total number of urinary catheter-days for all patients that have an indwelling urinary catheter in each location monitored
    - Standardization factor: Multiply by 1000 so that the measure is expressed as cases per 1000 catheter-days

b) Rate of bloodstream infections secondary to CAUTI: Use NHSN definitions for laboratory-confirmed bloodstream infection, available at http://www.cdc.gov/nhsn/library.html.
    - Numerator: number of episodes of bloodstream infections secondary to CAUTI
    - Denominator: total number of urinary catheter-days for all patients that have an indwelling urinary catheter in each location monitored
    - Standardization factor: Multiply by 1000 so that the measure is expressed as cases per 1000 catheter-days

B. External Reporting. Current NHSN definitions for CAUTI were developed for monitoring of rates within a facility; however, reporting of CAUTI rates for facility-to-facility comparison might be requested by state requirements and external quality initiatives.

IV. Recommendations for Further Research

Our literature review revealed that many of the studies addressing strategies to prevent CAUTI were not of sufficient quality to allow firm conclusions regarding the benefit of certain interventions. Future studies of CAUTI prevention should:

1) Be primary analytic research (i.e. systematic reviews, meta-analyses, interventional studies, and observational studies [cohort, case-control, analytic cross-sectional studies])
2) Evaluate clinically relevant outcomes (e.g., SUTI, bloodstream infections secondary to CAUTI)
3) Adjust for confounders as needed using multivariable analyses
4) Stratify outcomes by patient populations at risk for CAUTI
5) Ensure adequate statistical power to detect differences

The following is a compilation of recommendations for further research:

1. Catheter materials
    a. Antimicrobial and antiseptic-impregnated catheters
        i. Effect of catheters on reducing the risk of SUTI and other clinically significant outcomes
        ii. Patient populations most likely to benefit
        iii. Incidence of antimicrobial resistance in urinary pathogens
        iv. Role of bacterial biofilms in the pathogenesis of CAUTI
    b. Standard catheters
        i. Optimal materials for reducing the risk of CAUTI and other urethral complications

2. Appropriate urinary catheter use
    a. Incontinent patients
        i. Risks and benefits of periodic (e.g., nighttime) use of external catheters
        ii. Risk of local complications (e.g., skin maceration, phimosis) with the use of external catheters
        iii. Appropriate use of urinary catheters to manage sacral or perineal wounds
    b. Appropriate indications for continued use in postoperative patients and associated risks

3. Antiseptics
    a. Use of antiseptic vs. sterile solutions for periurethral cleaning prior to catheter insertion
    b. Use of antiseptics (e.g., methenamine) to prevent CAUTI

4. Alternatives to indwelling urethral catheters and bag drainage
    a. Risks and benefits of suprapubic catheters as an alternative to chronic indwelling urethral catheters
    b. Use of a urethral stent as an alternative to an indwelling catheter in selected patients with bladder outlet obstruction
    c. Use of catheter valves in reducing the risk of CAUTI and other urinary complications
    d. Other alternative methods of urinary drainage

5. Optimal methods for preventing encrustation in long-term catheterized patients who have frequent obstruction
    a. Optimal catheter materials
    b. Irrigation with acidifying solutions or oral urease inhibitors
    c. Use of methenamine

6. Other prevention measures
    a. Use of portable ultrasound in patients with low-urine output to reduce unnecessary catheter insertions or irrigations (in catheterized patients)
    b. Use of new prevention strategies such as bacterial interference in patients requiring chronic catheterization
    c. Optimal cleaning and storage procedures (e.g., wet vs. dry storage) for catheters used for clean intermittent catheterization

7. Prevention of transmission
    a. Spatial separation of patients with urinary catheters (in the absence of epidemic spread or frequent cross-infection) to prevent transmission of pathogens colonizing urinary drainage systems

V. Background

Urinary tract infections are the most common type of healthcare-associated infection, accounting for more than 30% of infections reported by acute care hospitals.[19] Virtually all healthcare-associated UTIs are caused by instrumentation of the urinary tract. Catheter-associated urinary tract infection (CAUTI) has been associated with increased morbidity, mortality, hospital cost, and length of stay.[6-9] In addition, bacteriuria commonly leads to unnecessary antimicrobial use, and urinary drainage systems are often reservoirs for multidrug-resistant bacteria and a source of transmission to other patients.[10,11]

Definitions

An indwelling urinary catheter is a drainage tube that is inserted into the urinary bladder through the urethra, is left in place, and is connected to a closed collection system. Alternative methods of urinary drainage may be employed in some patients. Intermittent ("in-and-out") catheterization involves brief insertion of a catheter into the bladder through the urethra to drain urine at intervals. An external catheter is a urine containment device that fits over or adheres to the genitalia and is attached to a urinary drainage bag. The most commonly used external catheter is a soft flexible sheath that fits over the penis ("condom" catheter). A suprapubic catheter is surgically inserted into the bladder through an incision above the pubis.

Although UTIs associated with alternative urinary drainage systems are considered device-associated, CAUTI rates reported to the National Healthcare Safety Network (NHSN) only refer to those associated with indwelling urinary catheters. NHSN has recently revised the UTI surveillance definition criteria. Among the changes are removal of the asymptomatic bacteriuria (ASB) criterion and refinement of the criteria for defining symptomatic UTI (SUTI). The time period for follow-up surveillance after catheter removal also has been shortened from 7 days to 48 hours to align with other device-associated infections. The new UTI criteria, which took effect in January 2009, can be found in the NHSN Patient Safety Manual (http://www.cdc.gov/nhsn/library.html).

The limitations and heterogeneity of definitions of CAUTI used in various studies present major challenges in appraising the quality of evidence in the CAUTI literature. Study investigators have used numerous different definitions for CAUTI outcomes, ranging from simple bacteriuria at a range of concentrations to, less commonly, symptomatic infection defined by combinations of bacteriuria and various signs and symptoms. Futhermore, most studies that used CDC/NHSN definitions for CAUTI did not distinguish between SUTI and ASB in their analyses.[30] The heterogeneity of definitions used for CAUTI may reduce the quality of evidence for a given intervention and often precludes meta-analyses.

The clinical significance of ASB in catheterized patients is undefined. Approximately 75% to 90% of patients with ASB do not develop a systemic inflammatory response or other signs or symptoms to suggest infection.[6,31] Monitoring and treatment of ASB is also not an effective prevention measure for SUTI, as most cases of SUTI are not preceded by bacteriuria for more than a day.[25] Treatment of ASB has not been shown to be clinically beneficial and is associated with the selection of antimicrobial-resistant organisms.

Epidemiology

Between 15% and 25% of hospitalized patients may receive short-term indwelling urinary catheters.[12,13] In many cases, catheters are placed for inappropriate indications, and healthcare providers are often unaware that their patients have catheters, leading to prolonged, unnecessary use.[14-16] In acute care hospitals reporting to NHSN in 2006, pooled mean urinary catheter utilization ratios in ICU and non-ICU areas ranged from 0.23-0.91 urinary catheter-days/patient-days.[17] While the numbers of units reporting were small, the highest ratios were in trauma ICUs and the lowest in inpatient medical/surgical wards. The overall prevalence of long-term indwelling urethral catheterization use is unknown. The prevalence of urinary catheter use in residents in long-term care facilities in the United States is on the order of 5%, representing approximately 50,000 residents with catheters at any given time.[18] This number appears to be declining over time, likely because of federally mandated nursing home quality measures. However, the high prevalence of urinary catheters in patients transferred to skilled nursing facilities suggests that acute care hospitals should focus more efforts on removing unnecessary catheters prior to transfer.[18]

Reported rates of UTI among patients with urinary catheters vary substantially. National data from NHSN acute care hospitals in 2006 showed a range of pooled mean CAUTI rates of 3.1-7.5 infections per 1000 catheter-days.[17] The highest rates were in burn ICUs, followed by inpatient medical wards and neurosurgical ICUs, although these sites also had the fewest numbers of locations reporting. The lowest rates were in medical/surgical ICUs.

Although morbidity and mortality from CAUTI is considered to be relatively low compared to other HAIs, the high prevalence of urinary catheter use leads to a large cumulative burden of infections with resulting infectious complications and deaths. An estimate of annual incidence of HAIs and mortality in 2002, based on a broad survey of US hospitals, found that urinary tract infections made up the highest number of infections (> 560,000) compared to other HAIs, and attributable deaths from UTI were estimated to be over 13,000 (mortality rate 2.3%).[19] And while fewer than 5% of bacteriuric cases develop bacteremia,[6] CAUTI is the leading cause of secondary nosocomial bloodstream infections; about 17% of hospital-acquired bacteremias are from a urinary source, with an associated mortality of approximately 10%.[20] In the nursing home setting, bacteremias are most commonly caused by UTIs, the majority of which are catheter-related.[21]

An estimated 17% to 69% of CAUTI may be preventable with recommended infection control measures, which means that up to 380,000 infections and 9000 deaths related to CAUTI per year could be prevented.[22]

Pathogenesis and Microbiology

The source of microorganisms causing CAUTI can be endogenous, typically via meatal, rectal, or vaginal colonization, or exogenous, such as via contaminated hands of healthcare personnel or equipment. Microbial pathogens can enter the urinary tract either by the extraluminal route, via migration along the outside of the catheter in the periurethral mucous sheath, or by the intraluminal route, via movement along the internal lumen of the catheter from a contaminated collection bag or catheter-drainage tube junction. The relative contribution of each route in the pathogenesis of CAUTI is not well known. The marked reduction in risk of bacteriuria with the introduction of the sterile, closed urinary drainage system in the1960's[23] suggests the importance of the intraluminal route. However, even with the closed drainage system,

bacteriuria inevitably occurs over time either via breaks in the sterile system or via the extraluminal route.[24] The daily risk of bacteriuria with catheterization is 3% to 10%,[25,26] approaching 100% after 30 days, which is considered the delineation between short and long-term catheterization.[27]

Formation of biofilms by urinary pathogens on the surface of the catheter and drainage system occurs universally with prolonged duration of catheterization.[28] Over time, the urinary catheter becomes colonized with microorganisms living in a sessile state within the biofilm, rendering them resistant to antimicrobials and host defenses and virtually impossible to eradicate without removing the catheter. The role of bacteria within biofilms in the pathogenesis of CAUTI is unknown and is an area requiring further research.

The most frequent pathogens associated with CAUTI (combining both ASB and SUTI) in hospitals reporting to NHSN between 2006-2007 were *Escherichia coli* (21.4%) and *Candida* spp (21.0%), followed by *Enterococcus* spp (14.9%), *Pseudomonas aeruginosa* (10.0%), *Klebsiella pneumoniae* (7.7%), and *Enterobacter* spp (4.1%). A smaller proportion was caused by other gram-negative bacteria and *Staphylococcus* spp [5].

Antimicrobial resistance among urinary pathogens is an ever increasing problem. About a quarter of *E. coli* isolates and one third of *P. aeruginosa* isolates from CAUTI cases were fluoroquinolone-resistant. Resistance of gram-negative pathogens to other agents, including third-generation cephalosporins and carbapenems, was also substantial [5]. The proportion of organisms that were multidrug-resistant, defined by non-susceptibility to all agents in 4 classes, was 4% of *P. aeruginosa*, 9% of *K. pneumoniae*, and 21% of *Acinetobacter baumannii*. [29]

VI. Scope and Purpose

This guideline updates and expands the original CDC Guideline for Prevention of CAUTI published in 1981. The revised guideline addresses the prevention of CAUTI for patients in need of either short- or long-term (i.e., > 30 days) urinary catheterization in any type of healthcare facility and evaluates evidence for alternative methods of urinary drainage, including intermittent catheterization, external catheters, and suprapubic catheters. The guideline also includes specific recommendations for implementation, performance measurement, and surveillance. Recommendations for further research are also provided to address the knowledge gaps in CAUTI prevention identified during the literature review.

To evaluate the evidence on preventing CAUTI, we examined data addressing three key questions and related subquestions:

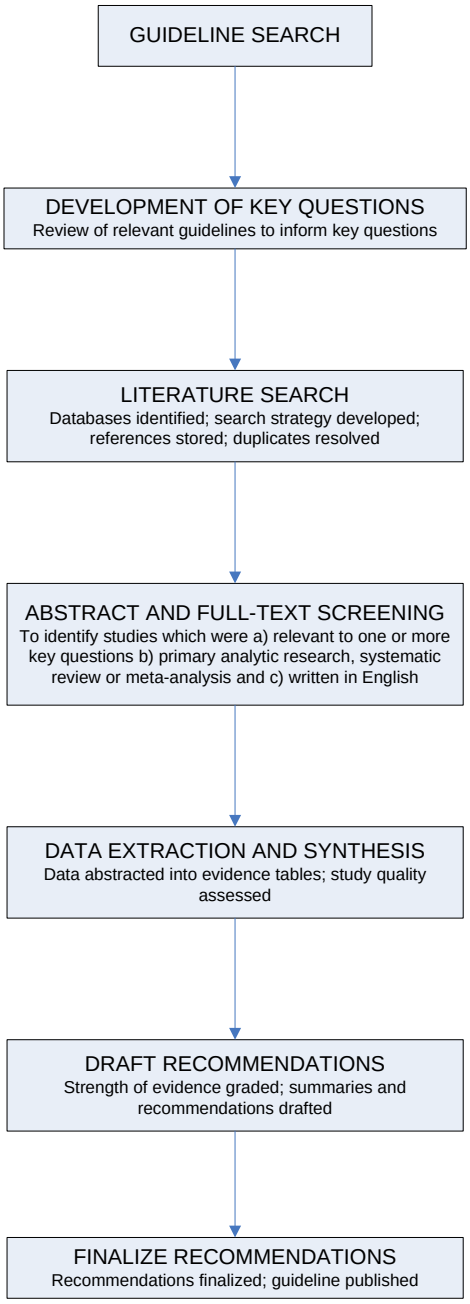
1. Who should receive urinary catheters?
    A. When is urinary catheterization necessary?
    B. What are the risk factors for CAUTI?
    C. What populations are at highest risk of mortality from catheters?
2. For those who may require urinary catheters, what are the best practices? Specifically, what are the risks and benefits associated with:
    A. Different approaches to catheterization?
    B. Different catheters or collecting systems?
    C. Different catheter management techniques?
    D. Different systems interventions (i.e., quality improvement programs)?
3. What are the best practices for preventing UTI associated with obstructed urinary catheters?

This document is intended for use by infection prevention staff, healthcare epidemiologists, healthcare administrators, nurses, other healthcare providers, and persons responsible for developing, implementing, and evaluating infection prevention and control programs for healthcare settings across the continuum of care. The guideline can also be used as a resource for societies or organizations that wish to develop more detailed implementation guidance for prevention of CAUTI.

VII. Methods

This guideline was based on a targeted systematic review of the best available evidence on CAUTI prevention. We used the Grading of Recommendations Assessment, Development and Evaluation (GRADE) approach [32-34] to provide explicit links between the available evidence and the resulting recommendations. Our guideline development process is outlined in *Figure 1.*

Figure 1. The Guideline Development Process



Development of Key Questions

We first conducted an electronic search of the National Guideline Clearinghouse® (Agency for Healthcare Research and Quality), Medline® (National Library of Medicine) using the Ovid® Platform (Ovid Technologies, Wolters Kluwer, New York, NY), the Cochrane® Health Technology Assessment Database (Cochrane Collaboration, Oxford, UK), the NIH Consensus Development Program, and the United States Preventive Services Task Force database for existing national and international guidelines relevant to CAUTI. The strategy used for the guideline search and the search results can be found in Appendix 1A. A preliminary list of key questions was developed from a review of the relevant guidelines identified in the search.[1,35,36] Key questions were finalized after vetting them with a panel of content experts and HICPAC members.

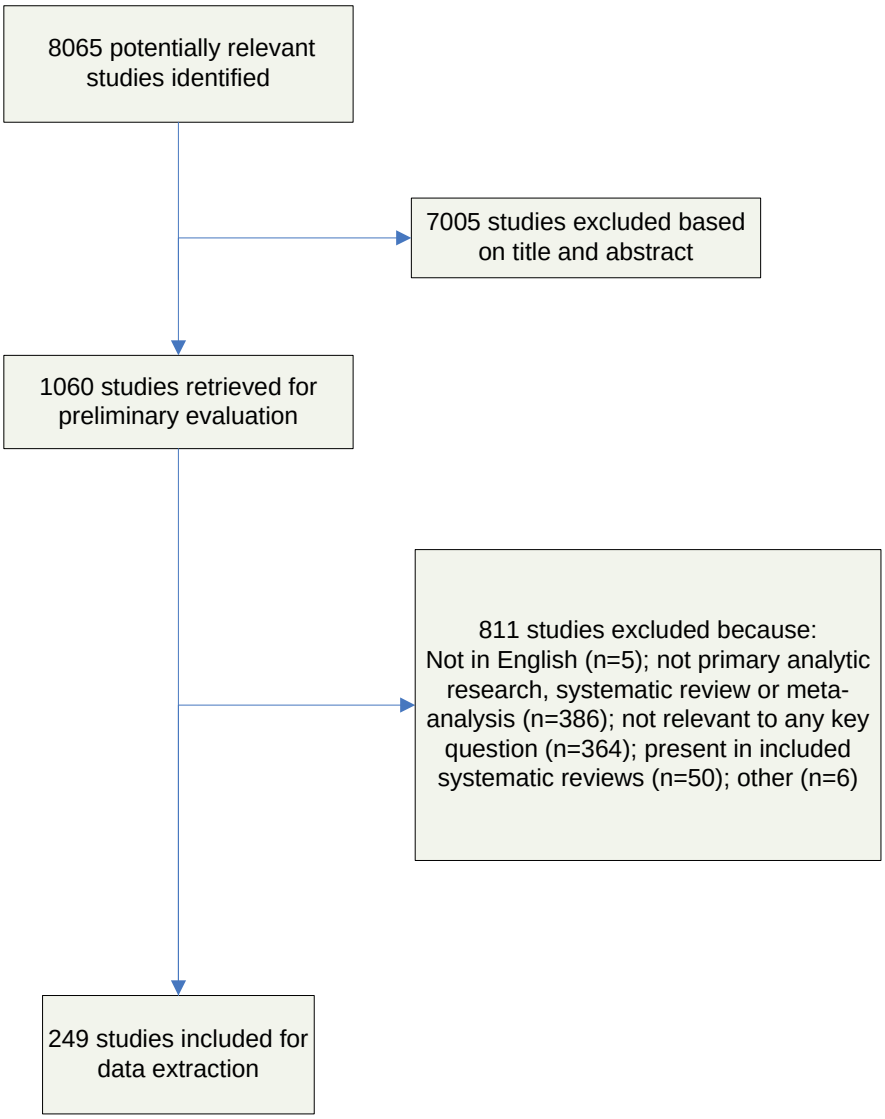
Literature Search

Following the development of the key questions, search terms were developed for identifying literature relevant to the key questions. For the purposes of quality assurance, we compared these terms to those used in relevant seminal studies and guidelines. These search terms were then incorporated into search strategies for the relevant electronic databases. Searches were performed in Medline® (National Library of Medicine) using the Ovid® Platform (Ovid Technologies, Wolters Kluwer, New York, NY), EMBASE® (Elsevier BV, Amsterdam, Netherlands), CINAHL® (Ebsco Publishing, Ipswich, MA) and Cochrane® (Cochrane Collaboration, Oxford, UK) (all databases were searched in July 2007), and the resulting references were imported into a reference manager, where duplicates were resolved. For Cochrane reviews ultimately included in our guideline, we checked for updates in July 2008. The detailed search strategy used for identifying primary literature and the results of the search can be found in Appendix 1B.

Study Selection

Titles and abstracts from references were screened by a single author (C.V.G, R.K.A., or D.A.P.) and the full text articles were retrieved if they were 1) relevant to one or more key questions, 2) primary analytic research, systematic reviews or meta-analyses, and 3) written in English. Likewise, the full-text articles were screened by a single author (C.V.G. or D.A.P.) using the same criteria, and included studies underwent a second review for inclusion by another author (R.K.A.). Disagreements were resolved by the remaining authors. The results of this process are depicted in *Figure 2.*

Figure 2: Results of the Study Selection Process



Data Extraction and Synthesis

Data on the study author, year, design, objective, population, setting, sample size, power, follow-up, and definitions and results of clinically relevant outcomes were extracted into evidence tables (Appendix 2). Three evidence tables were developed, each of which represented one of our key questions. Studies were extracted into the most relevant evidence table. Then, studies were organized by the common themes that emerged within each evidence table. Data were extracted by one author (R.K.A.) and cross-checked by another (C.V.G.). Disagreements were resolved by the remaining authors. Data and analyses were extracted as originally presented in the included studies. Meta-analyses were performed only where their use was deemed critical to a recommendation, and only in circumstances where multiple studies with sufficiently homogenous populations, interventions, and outcomes could be analyzed. Systematic reviews were included in our review. To avoid duplication of data, we excluded primary studies if they were also included in a systematic review captured by our search. The only exception to this was if the primary study also addressed a relevant question that was outside the scope of the included systematic review. Before exclusion, data from the primary studies that we originally captured were abstracted into the evidence tables and reviewed. We also excluded systematic reviews that analyzed primary studies that were fully captured in a more recent systematic review. The only exception to this was if the older systematic review also addressed a relevant question that was outside the scope of the newer systematic review. To ensure that all relevant studies were captured in the search, the bibliography was vetted by a panel of clinical experts.

Grading of Evidence

First, the quality of each study was assessed using scales adapted from existing methodology checklists, and scores were recorded in the evidence tables. Appendix 3 includes the sets of questions we used to assess the quality of each of the major study designs. Next, the quality of the evidence base was assessed using methods adapted from the GRADE Working Group.[32] Briefly, GRADE tables were developed for each of the interventions or questions addressed within the evidence tables. Included in the GRADE tables were the intervention of interest, any outcomes listed in the evidence tables that were judged to be clinically important, the quantity and type of evidence for each outcome, the relevant findings, and the GRADE of evidence for each outcome, as well as an overall GRADE of the evidence base for the given intervention or question. The initial GRADE of evidence for each outcome was deemed high if the evidence base included a randomized controlled trial (RCT) or a systematic review of RCTs, low if the evidence base included only observational studies, or very low if the evidence base consisted only of uncontrolled studies. The initial GRADE could then be modified by eight criteria.[34] Criteria which could decrease the GRADE of an evidence base included quality, consistency, directness, precision, and publication bias. Criteria that could increase the GRADE included a large magnitude of effect, a dose-response gradient, or inclusion of unmeasured confounders that would increase the magnitude of effect (Table 3). GRADE definitions are as follows:

1. High - further research is very unlikely to change confidence in the estimate of effect
2. Moderate - further research is likely to affect confidence in the estimate of effect and may change the estimate
3. Low - further research is very likely to affect confidence in the estimate of effect and is likely to change the estimate
4. Very low - any estimate of effect is very uncertain

After determining the GRADE of the evidence base for each outcome of a given intervention or question, we calculated the overall GRADE of the evidence base for that intervention or question. The overall GRADE was based on the lowest GRADE for the outcomes deemed critical to making a recommendation.

Table 3. Rating the Quality of Evidence Using the GRADE Approach

Type of Evidence	Initial Grade	Criteria to Decrease Grade	Criteria to Increase Grade	Overall Quality Grade
RCT	High	Quality Serious (-1 grade) or very serious (-2 grades) limitation to study quality Consistency Important inconsistency (-1 grade) Directness Some (-1 grade) or major (-2 grades) uncertainty about directness Precision Imprecise or sparse data (-1 grade) Publication bias High risk of bias (-1 grade)	Strong association Strong (+1 grade) or very strong evidence of association (+2 grades) Dose-response Evidence of a dose-response gradient (+1 grade) Unmeasured Confounders Inclusion of unmeasured confounders increases the magnitude of effect (+1 grade)	High Moderate Low Very low
Observational study	Low			
Any other evidence (e.g., expert opinion)	Very low			

Formulating Recommendations

Narrative evidence summaries were then drafted by the working group using the evidence and GRADE tables. One summary was written for each theme that emerged under each key question. The working group then used the narrative evidence summaries to develop guideline recommendations. Factors determining the strength of a recommendation included 1) the values and preferences used to determine which outcomes were "critical," 2) the harms and benefits that result from weighing the "critical" outcomes, and 3) the overall GRADE of the evidence base for the given intervention or question (Table 4).[33] If weighing the "critical outcomes" for a given intervention or question resulted in a "net benefit" or a "net harm," then a "Category I Recommendation" was formulated to strongly recommend for or against the given intervention respectively. If weighing the "critical outcomes" for a given intervention or question resulted in a "trade off" between benefits and harms, then a "Category II Recommendation" was formulated to recommend that providers or institutions consider the intervention when deemed appropriate. If weighing the "critical outcomes" for a given intervention or question resulted in

an "uncertain trade off" between benefits and harms, then a "No Recommendation" was formulated to reflect this uncertainty.

Table 4. Formulating Recommendations		
HICPAC Recommendation	**Weighing Benefits and Harms for Critical Outcomes**	**Quality of Evidence**
STRONG (I)	Interventions with net benefits or net harms	IA – High to Moderate IB – Low or Very Low (Accepted Practice) IC – High to Very Low (Regulatory)
WEAK (II)	Inteventions with trade offs between benefits and harms	High to Very Low
No recommendation/ unresolved issue	Uncertain trade offs between benefits and harms	Low to Very Low

For Category I recommendations, levels A and B represent the quality of the evidence underlying the recommendation, with A representing high to moderate quality evidence and B representing low quality evidence or, in the case of an established standard (e.g., aseptic technique, education and training), very low quality to no evidence based on our literature review. For IB recommendations, although there may be low to very low quality or even no available evidence directly supporting the benefits of the intervention, the theoretical benefits are clear, and the theoretical risks are marginal. Level C represents practices required by state or federal regulation, regardless of the quality of evidence. It is important to note that the strength of a Category IA recommendation is equivalent to that of a Category IB or IC recommendation; it is only the quality of the evidence underlying the IA recommendation that makes it different from a IB.

In some instances, multiple recommendations emerged from a single narrative evidence summary. The new HICPAC categorization scheme for recommendations is provided in Table 1, which is reproduced below.

Table 1. Modified HICPAC Categorization Scheme for Recommendations	
Category IA	A strong recommendation supported by high to moderate quality evidence suggesting net clinical benefits or harms
Category IB	A strong recommendation supported by low quality evidence suggesting net clinical benefits or harms or an accepted practice (e.g., aseptic technique) supported by low to very low quality evidence
Category IC	A strong recommendation required by state or federal regulation.
Category II	A weak recommendation supported by any quality evidence suggesting a trade off between clinical benefits and harms
No recommendation/ unresolved issue	Unresolved issue for which there is low to very low quality evidence with uncertain trade offs between benefits and harms

Category I recommendations are defined as strong recommendations with the following implications:

1. For patients: Most people in the patient's situation would want the recommended course of action and only a small proportion would not; request discussion if the intervention is not offered.
2. For clinicians: Most patients should receive the recommended course of action.
3. For policymakers: The recommendation may be adopted as a policy.

Category II recommendations are defined as weak recommendations with the following implications:

1. For patients: Most people in the patient's situation would want the recommended course of action, but many would not.
2. For clinicians: Different choices will be appropriate for different patients, and clinicians must help each patient to arrive at a management decision consistent with her or his values and preferences.
3. For policymakers: Policy making will require substantial debate and involvement of many stakeholders.

It should be noted that Category II recommendations are discretionary for the individual institution and are not intended to be enforced.

The wording of each recommendation was carefully selected to reflect the recommendation's strength. In most cases, we used the active voice when writing Category I recommendations - the strong recommendations. Phrases like "do" or "do not" and verbs without auxiliaries or conditionals were used to convey certainty. We used a more passive voice when writing Category II recommendations - the weak recommendations. Words like "consider" and phrases like "is preferable," "is suggested," "is not suggested," or "is not recommended" were chosen to reflect the lesser certainty of the Category II recommendations. Rather than a simple statement of fact, each recommendation is actionable, describing precisely a proposed action to take.

The category "No recommendation/unresolved issue" was most commonly applied to situations where either 1) the overall quality of the evidence base for a given intervention was low to very low and there was no consensus on the benefit of the intervention or 2) there was no published evidence on outcomes deemed critical to weighing the risks and benefits of a given intervention. If the latter was the case, those critical outcomes will be noted at the end of the relevant evidence summary.

Our evidence-based recommendations were cross-checked with those from guidelines identified in our original systematic search. Recommendations from previous guidelines for topics not directly addressed by our systematic review of the evidence were included in our "Summary of Recommendations" if they were deemed critical to the target users of this guideline. Unlike recommendations informed by our literature search, these recommendations are not linked to a key question. These recommendations were agreed upon by expert consensus and are designated either IB if they represent a strong recommendation based on accepted practices (e.g., aseptic technique) or II if they are a suggestion based on a probable net benefit despite limited evidence.
All recommendations were approved by HICPAC. Recommendations focused only on efficacy, effectiveness, and safety. The optimal use of these guidelines should include a consideration of the costs relevant to the local setting of guideline users.

Reviewing and Finalizing the Guideline

After a draft of the tables, narrative summaries, and recommendations was completed, the working group shared the draft with the expert panel for in-depth review. While the expert panel was reviewing this draft, the working group completed the remaining sections of the guideline, including the executive summary, background, scope and purpose, methods, summary of recommendations, and recommendations for guideline implementation, audit, and further research. The working group then made revisions to the draft based on feedback from members of the expert panel and presented the entire draft guideline to HICPAC for review. The guideline was then posted on the Federal Register for public comment. After a period of public comment, the guideline was revised accordingly, and the changes were reviewed and voted on by HICPAC. The final guideline was cleared internally by CDC and published and posted on the HICPAC website.

Updating the Guideline

Future revisions to this guideline will be dictated by new research and technological advancements for preventing CAUTI and will occur at the request of HICPAC.

VIII. Evidence Review

Q1. Who should receive urinary catheters?

To answer this question, we focused on three subquestions: A) When is urinary catheterization necessary? B) What are the risk factors for CAUTI? and C) What populations are at highest risk of mortality from urinary catheters?

Q1A. When is urinary catheterization necessary?

The available data examined five main populations. In all populations, we considered CAUTI outcomes as well as other outcomes we deemed critical to weighing the risks and benefits of catheterization. The evidence for this question consists of 1 systematic review,[37] 9 RCTs,[38-46] and 12 observational studies.[47-58] The findings of the evidence review and the grades for all important outcomes are shown in Evidence Review Table 1A.

For *operative patients*, low-quality evidence suggested a benefit of avoiding urinary catheterization.[37-44,47-49] This was based on a decreased risk of bacteriuria/unspecified UTI, no effect on bladder injury, and increased risk of urinary retention in patients without catheters. Urinary retention in patients without catheters was specifically seen following urogenital surgeries. The most common surgeries studied were urogenital, gynecological, laparoscopic, and orthopedic surgeries. Our search did not reveal data on the impact of catheterization on peri-operative hemodynamic management.

For *incontinent patients*, low-quality evidence suggested a benefit of avoiding urinary catheterization.[45,50-52] This was based on a decreased risk of both SUTI and bacteriuria/unspecified UTI in male nursing home residents without urinary catheters compared to those with continuous condom catheters. We found no difference in the risk of UTI between having a condom catheter only at night and having no catheter. Our search did not reveal data on the impact of catheterization on skin breakdown.

For *patients with bladder outlet obstruction*, very low-quality evidence suggested a benefit of a urethral stent over an indwelling catheter.[53] This was based on a reduced risk of bacteriuria in those receiving a urethral stent. Our search did not reveal data on the impact of catheterization versus stent placement on urinary complications.

For *patients with spinal cord injury*, very low-quality evidence suggested a benefit of avoiding indwelling urinary catheters.[54,56] This was based on a decreased risk of SUTI and bacteriuria in those without indwelling catheters (including patients managed with spontaneous voiding, clean intermittent catheterization [CIC], and external striated sphincterotomy with condom catheter drainage), as well as a lower risk of urinary complications, including hematuria, stones, and urethral injury (fistula, erosion, stricture).

For *children with myelomeningocele and neurogenic bladder*, very low-quality evidence suggested a benefit of CIC compared to urinary diversion or self voiding.[46,57,58] This was based on a decreased risk of bacteriuria/unspecified UTI in patients receiving CIC compared to urinary diversion, and a lower risk of urinary tract deterioration (defined by febrile urinary tract infection, vesicoureteral reflux, hydronephrosis, or increases in BUN or serum creatinine) compared to self-voiding and in those receiving CIC early (< 1 year of age) versus late (> 3 years of age).

Evidence Review Table 1A. When is urinary catheterization necessary?

1A.1. Use urinary catheters in operative patients only as necessary, rather than routinely. **(Category IB)**

1A.2. Avoid use of urinary catheters in patients and nursing home residents for management of incontinence. **(Category IB)**

1A.2.a. Further research is needed on periodic (e.g., nighttime) use of external catheters in incontinent patients or residents and the use of catheters to prevent skin breakdown. **(No recommendation/unresolved issue)**

1A.3. Further research is needed on the benefit of using a urethral stent as an alternative to an indwelling catheter in selected patients with bladder outlet obstruction. **(No recommendation/unresolved issue)**

1A.4. Consider alternatives to chronic indwelling catheters, such as intermittent catheterization, in spinal cord injury patients. **(Category II)**

1A.5. Consider intermittent catheterization in children with myelomeningocele and neurogenic bladder to reduce the risk of urinary tract deterioration. **(Category II)**

Q1B. What are the risk factors for CAUTI?

To answer this question, we reviewed the quality of evidence for those risk factors examined in more than one study. We considered the critical outcomes for decision-making to be SUTI and bacteriuria. The evidence for this question consists of 11 RCTs[59-69] and 37 observational studies.[9,50,54,70-103] The findings of the evidence review and the grades for all important outcomes are shown in Evidence Review Table 1B.

For *SUTI*, [50,54,61,62,74,75,79,83,102,103] low-quality evidence suggested that female sex, older age, prolonged catheterization, impaired immunity, and lack of antimicrobial exposure are risk factors. Very low quality evidence suggested that catheter blockage and low albumin level are also risk factors. For *bacteriuria*, [9,59-61,63-68,72,73,76-78,82,84-86,89-94,96-100] multiple risk factors were identified; there was high quality evidence for prolonged catheterization and moderate quality evidence for female sex, positive meatal cultures, and lack of antimicrobial exposure. Low-quality evidence also implicated the following risk factors for bacteriuria: older age, disconnection of the drainage system, diabetes, renal dysfunction, higher severity of illness, impaired immunity, placement of the catheter outside of the operating room, lower professional training of the person inserting the catheter, incontinence, and being on an orthopaedic or neurology service. Our search did not reveal data on adverse events and antimicrobial resistance associated with antimicrobial use, although one observational study found that the protective effect of antimicrobials lasted only for the first four days of catheterization, and that antimicrobial exposure led to changes in the epidemiology of bacterial flora in the urine.

Evidence Review Table 1B. What are the risk factors for CAUTI?

1B.1. Following aseptic insertion of the urinary catheter, maintain a closed drainage system. **(Category IB)**[a]

1B.2. Insert catheters only for appropriate indications, and leave in place only as long as needed. **(Category IB)**[b]

1B.3. Minimize urinary catheter use and duration of use in all patients, particularly those at higher risk for CAUTI such as women, the elderly, and patients with impaired immunity. **(Category IB)**

1B.4. Ensure that only properly trained persons (e.g., hospital personnel, family members, or patients themselves) who know the correct technique of aseptic catheter insertion and maintenance are given this responsibility. **(Category IB)**

1B.5. Maintain unobstructed urine flow. **(Category IB)**[c]

[a] More data are available under Question 2B.
[b] More data are available under Question 2C.
[c] More data are available under Question 2D.

Q1C. What populations are at highest risk of mortality from urinary catheters?

To answer this question, we reviewed the quality of evidence for those risk factors examined in more than one study. The evidence for this question consists of 2 observational studies.[7,74] The findings of the evidence review and the grades for all important outcomes are shown in Evidence Review Table 1C.

Low-quality evidence suggested that older age, higher severity of illness, and being on an internal medicine service compared to a surgical service were independent risk factors for mortality in patients with indwelling urinary catheters. Both studies evaluating these risk factors found the highest risk of mortality in patients over 70 years of age. Low-quality evidence also suggested that CAUTI was a risk factor for mortality in patients with catheters.

Evidence Review Table 1C. What populations are at highest risk of mortality from catheters?

1C.1. Minimize urinary catheter use and duration in all patients, particularly those who may be at higher risk for mortality due to catheterization, such as the elderly and patients with severe illness. **(Category IB)**

Q2. For those who may require urinary catheters, what are the best practices?

To answer this question, we focused on four subquestions: A) What are the risks and benefits associated with different approaches to catheterization?, B) What are the risks and benefits associated with different types of catheters or collecting systems?, C) What are the risks and benefits associated with different catheter management techniques, and D) What are the risks and benefits associated with different systems interventions?

Q2A. What are the risks and benefits associated with different approaches to catheterization?

The available data examined the following comparisons of different catheterization approaches:

1) External versus indwelling urethral
2) Intermittent versus indwelling urethral
3) Intermittent versus suprapubic
4) Suprapubic versus indwelling urethral
5) Clean intermittent versus sterile intermittent

For all comparisons, we considered SUTI, bacteriuria/unspecified UTI, or combinations of these outcomes depending on availability, as well as other outcomes critical to weighing the risks and benefits of different catheterization approaches. The evidence for this question consists of 6 systematic reviews,[37,104-108] 16 RCTs,[62,63,109-122] and 18 observational studies.[54,73,81,84,123-136] The findings of the evidence review and the grades for all important outcomes are shown in Evidence Review Table 2A

Q2A.1. External versus indwelling urethral

Low-quality evidence suggested a benefit of using external catheters over indwelling urethral catheters in male patients who require a urinary collection device but do not have an indication for an indwelling catheter such as urinary retention or bladder outlet obstruction.[81,109,123] This was based on a decreased risk of a composite outcome of SUTI, bacteriuria, or death as well as increased patient satisfaction with condom catheters. Differences were most pronounced in men without dementia. Statistically significant differences were not found or reported for the individual CAUTI outcomes or death. Our search did not reveal data on differences in local complications such as skin maceration or phimosis.

Q2A.2. Intermittent versus indwelling urethral

Low-quality evidence suggested a benefit of using intermittent catheterization over indwelling urethral catheters in selected populations.[84,104-106,110-114,124-126,135,136] This was based on a decreased risk of SUTI and bacteriuria/unspecified UTI but an increased risk of urinary retention in postoperative patients with intermittent catheterization. In one study, urinary retention and bladder distension were avoided by performing catheterization at regular intervals (every 6-8 hrs) until return of voiding. Studies of patients with neurogenic bladder most consistently found a decreased risk of CAUTI with intermittent catheterization. Studies in operative patients whose catheters were removed within 24 hrs of surgery found no differences in bacteriuria with intermittent vs. indwelling catheterization, while studies where catheters were left in for longer durations had mixed results. Our search did not reveal data on differences in patient satisfaction.

Q2A.3. Intermittent versus suprapubic

Very low-quality evidence suggested a benefit of intermittent over suprapubic catheterization in selected populations[115,116,134-136] based on increased patient acceptability and decreased risk of urinary complications (bladder calculi, vesicoureteral reflux, and upper tract abnormalities). Although we found a decreased risk of bacteriuria/unspecified UTI with suprapubic catheterization, there were no differences in SUTI. The populations studied included women undergoing urogynecologic surgery and spinal cord injury patients.

Q2A.4. Suprapubic versus indwelling urethral

Low-quality evidence suggested a benefit of suprapubic catheters over indwelling urethral catheters in selected populations.[37,62,104,107,108,128-133,135,136] This was based on a decreased risk of bacteriuria/unspecified UTI, recatheterization, and urethral stricture, and increased patient comfort and satisfaction. However, there were no differences in SUTI and an increased risk of longer duration of catheterization with suprapubic catheters. Studies involved primarily postoperative and spinal cord injury patients. Our search did not reveal data on differences in complications related to catheter insertion or the catheter site.

Q2A.5. Clean intermittent versus sterile intermittent

Moderate-quality evidence suggested no benefit of using sterile over clean technique for intermittent catheterization.[63,73,105,117-122] No differences were found in the risk of SUTI or bacteriuria/unspecified UTI. Study populations included nursing home residents and adults and children with neurogenic bladder/spinal cord injury.

Evidence Review Table 2A. What are the risks and benefits associated with different approaches to catheterization?

2A.1. Consider using external catheters as an alternative to indwelling urethral catheters in cooperative male patients without urinary retention or bladder outlet obstruction. **(Category II)**

2A.2. Intermittent catheterization is preferable to indwelling urethral or suprapubic catheters in patients with bladder emptying dysfunction. **(Category II)**

2A.3. If intermittent catheterization is used, perform it at regular intervals to prevent bladder overdistension. **(Category IB)**

2A.4. For operative patients who have an indication for an indwelling catheter, remove the catheter as soon as possible postoperatively, preferably within 24 hours, unless there are appropriate indications for continued use. **(Category IB)***

2A.5. Further research is needed on the risks and benefits of suprapubic catheters as an alternative to indwelling urethral catheters in selected patients requiring short- or long-term catheterization, particularly with respect to complications related to catheter insertion or the catheter site. **(No recommendation/unresolved issue)**

2A.6. In the non-acute care setting, clean (i.e., non-sterile) technique for intermittent catheterization is an acceptable and more practical alternative to sterile technique for patients requiring chronic intermittent catheterization. **(Category IA)**

* More data are available under Question 2C

Q2B. What are the risks and benefits associated with different catheters or collecting systems?

The available data examined the following comparisons between different types of catheters and drainage systems:

1. Antimicrobial/antiseptic catheters vs. standard catheters
    a. Silver-coated catheters vs. standard catheters
    b. Nitrofurazone-impregnated catheters vs. standard catheters
2. Hydrophilic catheters vs. standard catheters
3. Closed vs. open drainage systems
4. Complex vs. simple drainage systems
5. Preconnected/sealed junction catheters vs. standard catheters
6. Catheter valves vs. catheter bags

For all comparisons, we considered CAUTI outcomes as well as other outcomes critical to weighing the risks and benefits of different types of catheters or collecting systems. The evidence for this question consists of 5 systematic reviews,[37,137-140] 17 RCTs,[64,143-158] 23 observational studies,[82,86,89,97,159-163, 165-178] and 3 economic analyses.[179180,181] The findings of the evidence review and the grades for all important outcomes are shown in Evidence Review Table 2B.

Q2B.1.a. Silver-coated catheters vs. standard catheters

Low-quality evidence suggested a benefit of silver-coated catheters over standard latex catheters.[37,82,86,137-139,143,159-163, 165,166] This was based on a decreased risk of bacteriuria/unspecified UTI with silver-coated catheters and no evidence of increased urethral irritation or antimicrobial resistance in studies that reported data on microbiological outcomes. Differences were significant for silver alloy-coated catheters but not silver oxide-coated catheters. In a meta-analysis of randomized controlled trials (see Appendix), silver alloy-coated catheters reduced the risk of asymptomatic bacteriuria compared to standard latex catheters (control latex catheters were either uncoated or coated with hydrogel, Teflon®, or silicone), whereas there were no differences when compared to standard, all silicone catheters. The effect of silver alloy catheters compared to latex catheters was more pronounced when used in patients catheterized <1 week. The results were robust to inclusion or exclusion of non peer-reviewed studies. Only one observational study found a decrease in SUTI with silver alloy-coated catheters.[166] The setting was a burn referral center, where the control catheters were latex, and patients in the intervention group had new catheters placed on admission, whereas the control group did not. Recent observational studies in hospitalized patients found mixed results for bacteriuria/unspecified UTI.

Q2B.1.b. Nitrofurazone-impregnated catheters vs. standard catheters

Low-quality evidence suggested a benefit of nitrofurazone-impregnated catheters in patients catheterized for short periods of time.[137,138] This was based on a decreased risk of bacteriuria and no evidence of increased antimicrobial resistance in studies that reported microbiological outcomes. Differences were significant in a meta-analysis of three studies examining nitrofurazone-impregnated catheters (only one individual study significant) when duration of catheterization was <1 week. No differences were seen when duration of catheterization was >1 week, although the meta-analysis was borderline significant.

Q2B.2. Hydrophilic catheters vs. standard catheters

Very low-quality evidence suggested a benefit of hydrophilic catheters over standard non-hydrophilic catheters in specific populations undergoing clean intermittent catheterization.[137,144-148,169] This was based on a decreased risk of SUTI, bacteriuria, hematuria, and pain during insertion, and increased patient satisfaction. Differences in CAUTI outcomes were limited to one study of spinal cord injury patients and one study of patients receiving intravesical immunochemoprophylaxis for bladder cancer, while multiple other studies found no significant differences.

Q2B.3. Closed vs. open drainage systems

Very low-quality evidence suggested a benefit of using a closed rather than open urinary drainage system.[89,171] This was based on a decreased risk of bacteriuria with a closed drainage system. One study also found a suggestion of a decreased risk of SUTI, bacteremia, and UTI-related mortality associated with closed drainage systems, but differences were not statistically significant. Sterile, continuously closed drainage systems became the standard of care based on an uncontrolled study published in 1966 demonstrating a dramatic reduction in the risk of infection in short-term catheterized patients with the use of a closed system.[23] Recent data also include the finding that disconnection of the drainage system is a risk factor for bacteriuria (Q1B).

Q2B.4. Complex vs. simple drainage systems

Low-quality evidence suggested no benefit of complex closed urinary drainage systems over simple closed urinary drainage systems.[150-152,154,172,176,177] Although there was a decreased risk of bacteriuria with the complex systems, differences were found only in studies published before 1990, and not in more recent studies. The complex drainage systems studied included various mechanisms for reducing bacterial entry, such as antiseptic-releasing cartridges at the drain port of the urine collection bag; see evidence table for systems evaluated.

Q2B.5. Preconnected/sealed junction catheters vs. standard catheters

Low-quality evidence suggested a benefit of using preconnected catheters with junction seals over catheters with unsealed junctions to reduce the risk of disconnections.[64,153,156,175] This was based on a decreased risk of SUTI and bacteriuria with preconnected sealed catheters. Studies that found differences had higher rates of CAUTI in the control group than studies that did not find an effect.

Q2B.6. Catheter valves vs. drainage bags

Moderate-quality evidence suggested a benefit of catheter valves over drainage bags in selected patients with indwelling urinary catheters.[140] Catheter valves led to greater patient satisfaction but no differences in bacteriuria/unspecified UTI or pain/bladder spasms. Details regarding the setting for recruitment and follow-up of the patients in the studies were unclear, and the majority of subjects were men. Our search did not reveal data on the effect of catheter valves on bladder function, bladder/urethral trauma, or catheter blockage.

Evidence Review Table 2B. What are the risks and benefits associated with different catheters or collecting systems?

2B.1. If the CAUTI rate is not decreasing after implementing a comprehensive strategy to reduce rates of CAUTI, consider using antimicrobial/antiseptic-impregnated catheters. The comprehensive strategy should include, at a minimum, the high priority recommendations for urinary catheter use, aseptic insertion, and maintenance (see Section III. Implementation and Audit). **(Category IB)**

2B.1.a. Further research is needed on the effect of antimicrobial/antiseptic-impregnated catheters in reducing the risk of symptomatic UTI, their inclusion among the primary interventions, and the patient populations most likely to benefit from these catheters. **(No recommendation/unresolved issue)**

2B.2. Hydrophilic catheters might be preferable to standard catheters for patients requiring intermittent catheterization. **(Category II)**

2B.3. Following aseptic insertion of the urinary catheter, maintain a closed drainage system. **(Category IB)**

2B.4. Complex urinary drainage systems (utilizing mechanisms for reducing bacterial entry such as antiseptic-release cartridges in the drain port) are not necessary for routine use. **(Category II)**

2B.5. Urinary catheter systems with preconnected, sealed catheter-tubing junctions are suggested for use. **(Category II)**

2B.6. Further research is needed to clarify the benefit of catheter valves in reducing the risk of CAUTI and other urinary complications. **(No recommendation/unresolved issue)**

Q2C. What are the risks and benefits associated with different catheter management techniques?

The available data examined the following catheter management techniques:

1. Antimicrobial prophylaxis
2. Urinary antiseptics (i.e., methanamine)
3. Bladder irrigation
4. Antiseptic instillation in the drainage bag
5. Periurethral care
6. Routine catheter or bag change
7. Catheter lubricants
8. Securing devices
9. Bacterial interference
10. Catheter cleansing
11. Catheter removal strategies (clamping vs. free drainage prior to removal, postoperative duration of catheterization)
12. Assessment of urine volumes

For all comparisons, we considered CAUTI outcomes as well as other outcomes critical to weighing the risks and benefits of different catheter management techniques. The evidence for this question consists of 6 systematic reviews,[37,105,106,182-184] 56 RCTs,[60,61,65-69,143,158,158,185-231] 34 observational studies,[83,85,88,90,96,102,133,167,178,232-258] and 1 economic analysis.[180] The findings of the evidence review and the grades for all important outcomes are shown in Evidence Review Table 2C.

Q2C.1. Antimicrobial prophylaxis

Low-quality evidence suggested no benefit of antimicrobial prophylaxis in patients undergoing short-term catheterization.[37,60,61,83,85,133,158,178,182,185,186,189-191,232-234] This was based on heterogeneous results for SUTI and bacteriuria/unspecified UTI and no adverse events related to antimicrobials. Lack of consistency in specific factors, such as patient population, antimicrobial agents, timing of administration, and duration of follow-up, did not allow for a summary of evidence of the effect of antimicrobial prophylaxis on CAUTI in patients undergoing short term catheterization. Only two studies evaluated adverse events related to antimicrobials. Our search did not reveal data on antimicrobial resistance or *Clostridium difficile* infection.

Low-quality evidence suggested no benefit of antimicrobial prophylaxis in patients undergoing long-term catheterization (indwelling and clean intermittent catheterization).[106,183,192,194,235,238] This was based on a decreased risk of bacteriuria, heterogeneous results for SUTI, and no differences reported for catheter encrustation or adverse events, although data were sparse. One systematic review suggested an increase in antimicrobial resistance with antimicrobial use.

Q2C.2. Urinary antiseptics

Low-quality evidence suggested a benefit of methenamine for short-term catheterized patients.[196,197] This was based on a reduced risk of SUTI and bacteriuria and no differences in adverse events. Evidence was limited to two studies of patients following gynecological surgery in Norway and Sweden.

Very low-quality evidence suggested a benefit of methanamine for long-term catheterized patients.[106,236-239] This was based on a reduced risk of encrustation but no differences in risk of SUTI or bacteriuria. Data on encrustation was limited to one study. Studies involved primarily elderly and spinal cord injury patients with chronic indwelling catheters

Q2C.3. Bladder irrigation

Low-quality evidence suggested no benefit of bladder irrigation in patients with indwelling or intermittent catheters.[66,69,199-206,240-242] This was based on no differences in SUTI and heterogeneous findings for bacteriuria.

Q2C.4. Antiseptic instillation in the drainage bag

Low-quality evidence suggested no benefit of antiseptic instillation in urinary drainage bags.[90,207-211,243-245] This was based on no differences in SUTI and heterogeneous results for bacteriuria.

Q2C.5. Periurethral care

Low-quality evidence suggested no benefit of antiseptic meatal cleaning regimens before or during catheterization to prevent CAUTI.[65,67,68,88,158,212-216,246,247] This was based on no difference in the risk of bacteriuria in patients receiving periurethral care regimens compared to those not receiving them. One study found a higher risk of bacteriuria with cleaning of the urethral meatus-catheter junction (either twice daily application of povidine-iodine or once daily cleaning with a non-antiseptic solution of green soap and water) in a subgroup of women with positive meatal cultures and in patients not receiving antimicrobials. Periurethral cleaning with chlorhexidine before catheter insertion did not have an effect in two studies.

Q2C.6. Routine catheter or bag change

Low-quality evidence suggested no benefit of routine catheter or drainage bag changes to prevent CAUTI.[102,217-219,248,249] This was based on no difference or an increased risk of SUTI and no difference in bacteriuria with routine compared to as-needed changes or with more frequent changing intervals. One study in nursing home residents found no differences in SUTI with routine monthly catheter changes compared to changing only for obstruction or infection, but the study was underpowered to detect a difference. Another study in home care patients found an increased risk of SUTI when catheters were changed more frequently than monthly.

Q2C.7. Catheter lubricants

Very low-quality evidence suggested a benefit of using lubricants for catheter insertion.[167,220-223,250-254] This was based on a decreased risk of SUTI and bacteriuria with the use of a pre-lubricated catheter compared to a catheter lubricated by the patient and a decreased risk of bacteriuria with use of a lubricant versus no lubricant. Studies were heterogeneous both in the interventions and outcomes studied. Several studies comparing antiseptic lubricants to non-antiseptic lubricants found no significant differences.

Q2C.8. Securing devices

Low-quality evidence suggested no benefit of using catheter securing devices to prevent CAUTI.[224] This was based on no significant difference in the risk of SUTI or meatal erosion. The only study in this category looked at one particular product.

Q2C.9. Bacterial interference

Moderate-quality evidence suggested a benefit of using bacterial interference in catheterized patients.[225] In the one study evaluating this intervention, urinary colonization with a non-pathogenic *Escherichia coli* was associated with a decreased risk of SUTI in adults with spinal cord injury and a history of frequent CAUTI.

Q2C.10. Catheter cleansing

Very low-quality evidence suggested a benefit of wet versus dry storage procedures for catheters used in clean intermittent catheterization.[255] This was based on a decreased risk of SUTI with a wet storage procedure in one study of spinal cord injury patients undergoing clean intermittent catheterization compared to a dry storage procedure where the catheter was left to air dry after washing. In the wet procedure, the catheter was stored in a dilute povidone-iodine solution after washing with soap and water.

Q2C.11. Catheter removal strategies

a. Clamping vs. free drainage prior to removal

Low-quality evidence suggested no benefit of clamping versus free drainage before catheter removal.[37,184] This was based on no difference in risk of bacteriuria, urinary retention, or recatheterization between the two strategies. One study comparing a clamp and release strategy to free drainage over 72 hours found a greater risk of bacteriuria in the clamping group.

b. Postoperative duration of catheterization

Moderate-quality evidence suggested a benefit of shorter versus longer postoperative durations of catheterization.[37,184,227,228] This was based on a decreased risk of bacteriuria/unspecified UTI, decreased time to ambulation and length of stay, no differences in urinary retention and SUTI, and increased risk of recatheterization. Significant decreases in bacteriuria/unspecified UTI were found specifically for comparisons of 1 day versus 3 or 5 days of postoperative catheterization. Recatheterization risk was greater in only one study comparing immediate removal to removal 6 or 12 hours after hysterectomy.

Q2C.12. Assessment of urine volumes

Low-quality evidence suggested a benefit of using portable ultrasound to assess urine volume in patients undergoing intermittent catheterization.[229,230] This was based on fewer catheterizations but no reported differences in risk of unspecified UTI. Patients studied were adults with neurogenic bladder in inpatient rehabilitation centers. Our search did not reveal data on the use of ultrasound in catheterized patients in other settings.

Evidence Review Table 2C. What are the risks and benefits associated with different catheter management techniques?

2C.1. Unless clinical indications exist (e.g., in patients with bacteriuria upon catheter removal post urologic surgery), do not use systemic antimicrobials routinely as prophylaxis for UTI in patients requiring either short or long-term catheterization. **(Category IB)**

2C.2.a. Further research is needed on the use of urinary antiseptics (e.g., methanamine) to prevent UTI in patients requiring short-term catheterization. **(No recommendation/unresolved issue)**
2C.2.b. Further research is needed on the use of methanamine to prevent encrustation in patients requiring chronic indwelling catheters who are at high risk for obstruction. **(No recommendation/unresolved issue)**

2C.3.a. Unless obstruction is anticipated (e.g., as might occur with bleeding after prostatic or bladder surgery), bladder irrigation is not recommended. **(Category II)**
2C.3.b. Routine irrigation of the bladder with antimicrobials is not recommended. **(Category II)**

2C.4. Routine instillation of antiseptic or antimicrobial solutions into urinary drainage bags is not recommended. **(Category II)**

2C.5.a. Do not clean the periurethral area with antiseptics to prevent CAUTI while the catheter is in place. Routine hygiene (e.g., cleansing of the meatal surface during daily bathing) is

appropriate. **(Category IB)**
2C.5.b. Further research is needed on the use of antiseptic solutions vs. sterile water or saline for periurethral cleaning prior to catheter insertion. **(No recommendation/unresolved issue)**

2C.6. Changing indwelling catheters or drainage bags at routine, fixed intervals is not recommended. Rather, catheters and drainage bags should be changed based on clinical indications such as infection, obstruction, or when the closed system is compromised. **(Category II)**

2C.7.a. Use a sterile, single-use packet of lubricant jelly for catheter insertion. **(Category IB)**
2C.7.b. Routine use of antiseptic lubricants is not necessary. **(Category II)**

2C.8. Further research is needed on the use of bacterial interference to prevent UTI in patients requiring chronic urinary catheterization. **(No recommendation/unresolved issue)**

2C.9. Further research is needed on optimal cleaning and storage methods for catheters used for clean intermittent catheterization. **(No recommendation/unresolved issue)**

2C.10.a. Clamping indwelling catheters prior to removal is not necessary. **(Category II)**
2C.10.b. Insert catheters only for appropriate indications, and leave in place only as long as needed. **(Category IB)**
2C.10.c. For operative patients who have an indication for an indwelling catheter, remove the catheter as soon as possible postoperatively, preferably within 24 hours, unless there are appropriate indications for continued use. **(Category IB)**

2C.11.a. Consider using a portable ultrasound device to assess urine volume in patients undergoing intermittent catheterization to assess urine volume and reduce unnecessary catheter insertions. **(Category II)**
2C.11.b. Further research is needed on the use of a portable ultrasound device to evaluate for obstruction in patients with indwelling catheters and low urine output. **(No recommendation/unresolved issue)**

Q2D. What are the risks and benefits associated with different systems interventions?

The available data examined the following systems interventions:

1. Infection control/quality improvement programs (multifaceted)
2. Catheter reminders
3. Bacteriologic monitoring
4. Hand hygiene
5. Patient placement
6. Catheter team versus self-catheterization
7. Feedback
8. Nurse-directed catheter removal

We considered CAUTI outcomes, duration of catheterization, recatheterization, and transmission of pathogens when weighing the risks and benefits of different systems interventions. The evidence for this question consists of 1 RCT[259] and 19 observational

studies.[3,25,260-276] The findings of the evidence review and the grades for all important outcomes are shown in Evidence Review Table 2D.

Q2D.1. Multifaceted infection control/quality improvement programs

Low-quality evidence suggested a benefit of multifaceted infection control/quality improvement programs to reduce the risk of CAUTI.[3,260-267] This was based on a decreased risk of SUTI, bacteriuria/unspecified UTI, and duration of catheter use with implementation of such programs. Studies evaluated various multifaceted interventions. The studies with significant findings included: 1) education and performance feedback regarding compliance with catheter care, emphasizing hand hygiene, and maintaining unobstructed urine flow; 2) computerized alerts to physicians, nurse-driven protocols to remove catheters, and use of handheld bladder scanners to assess for urinary retention; 3) guidelines and education focusing on perioperative catheter management; and 4) a multifaceted infection control program including guidelines for catheter insertion and maintenance. A program using a checklist and algorithm for appropriate catheter use also suggested a decrease in unspecified UTI and catheter duration, but statistical differences were not reported.

Q2D.2. Reminders

Very low-quality evidence suggested a benefit of using urinary catheter reminders to prevent CAUTI.[268-270] This was based on a decreased risk of bacteriuria and duration of catheterization and no differences in recatheterization or SUTI when reminders were used. Reminders to physicians included both computerized and non-computerized alerts about the presence of urinary catheters and the need to remove unnecessary catheters.

Q2D.3. Bacteriologic monitoring

Very low-quality evidence suggested no benefit of bacteriologic monitoring to prevent CAUTI.[25,271] Although one study found a decreased risk of bacteriuria during a period of bacteriologic monitoring and feedback, only 2% of SUTI episodes were considered potentially preventable with the use of bacteriologic monitoring.

Q2D.4. Hand hygiene

Very low-quality evidence suggested a benefit of using alcohol hand sanitizer in reducing CAUTI. This was based on one study in a rehabilitation facility that found a decrease in unspecified UTI, although no statistical differences were reported.[272] A separate multifaceted study that included education and performance feedback on compliance with catheter care and hand hygiene showed a decrease in risk of SUTI.[265]

Q2D.5. Patient placement

Very low-quality evidence suggested a benefit of spatially separating patients to prevent transmission of urinary pathogens.[273] This was based on a decreased risk of transmission of urinary bacterial pathogens in nursing home residents in separate rooms compared to residents in the same rooms.

Q2D.6. Catheter team versus self-catheterization

Very low-quality evidence suggested no benefit of a catheter team to prevent CAUTI among patients requiring intermittent catheterization.[274] This was based on one study showing no difference in unspecified UTI between use of a catheter care team and self-catheterization for intermittent catheterization in paraplegic patients.

Q2D.7. Feedback

Very low-quality evidence suggested a benefit of using nursing feedback to prevent CAUTI.[275] This was based on a decreased risk of unspecified UTI during an intervention where nursing staff were provided with regular reports of unit-specific rates of CAUTI.

Q2D.8. Nurse-directed catheter removal

Very low-quality evidence suggested a benefit of a nurse-directed catheter removal program to prevent CAUTI.[276] This was based on a decreased risk of unspecified UTI during an intervention where criteria were developed that allowed a registered nurse to remove a catheter without a physician's order when no longer medically necessary. Of the three intensive care units where the intervention was implemented, differences were significant only in the coronary intensive care unit.

Evidence Review Table 2D. What are the risks and benefits associated with different systems interventions?

2D.1.a. Ensure that healthcare personnel and others who take care of catheters are given periodic in-service training stressing the correct techniques and procedures for urinary catheter insertion, maintenance, and removal. **(Category IB)**
2D.1.b. Implement quality improvement (QI) programs or strategies to enhance appropriate use of indwelling catheters and to reduce the risk of CAUTI based on a facility risk assessment. **(Category IB)**

Examples of programs that have been demonstrated to be effective include:

1. A system of alerts or reminders to identify all patients with urinary catheters and assess the need for continued catheterization
2. Guidelines and protocols for nurse-directed removal of unnecessary urinary catheters
3. Education and performance feedback regarding appropriate use, hand hygiene, and catheter care
4. Guidelines and algorithms for appropriate peri-operative catheter management, such as:
    a. Procedure-specific guidelines for catheter placement and postoperative catheter removal
    b. Protocols for management of postoperative urinary retention, such as nurse-directed use of intermittent catheterization and use of ultrasound bladder scanners

2D.2. Routine screening of catheterized patients for asymptomatic bacteriuria is not recommended. **(Category II)**

2D.3. Perform hand hygiene immediately before and after insertion or any manipulation of the catheter site or device. **(Category IB)**

2D.5. Maintain unobstructed urine flow. **(Category IB)**

2D.6. Further research is needed on the benefit of spatial separation of patients with urinary catheters to prevent transmission of pathogens colonizing urinary drainage systems. **(No recommendation/unresolved issue)**

2D.7. When performing surveillance for CAUTI, consider providing regular (e.g., quarterly) feedback of unit-specific CAUTI rates to nursing staff and other appropriate clinical care staff. **(Category II)**

Q3: What are the best practices for preventing UTI associated with obstructed urinary catheters?

The available data examined the following practices:

1. Methods to prevent/reduce encrustations or blockage
2. Catheter materials preventing blockage

For this question, available relevant outcomes included blockage/encrustation. We did not find data on the outcomes of CAUTI. The evidence for this question consists of 1 systematic review,[277] 2 RCTs,[278,279] and 2 observational studies.[280,281] The findings of the evidence review and the grades for all important outcomes are shown in Evidence Review Table 3.

Q3.1. Methods to prevent/reduce encrustations or blockage

Low-quality evidence suggested a benefit of acidifying solutions or oral acetohydroxamic acid in preventing or reducing catheter encrustations and blockage in long-term catheterized patients.[277,278,280,281] No differences were seen with daily catheter irrigation with normal saline.

Q3.2. Catheter materials preventing blockage

Low-quality evidence suggested a benefit of silicone over latex or Teflon-coated catheters in prevention or reducing catheter encrustations in long-term catheterized patients who were prone to blockage. No differences were seen with different materials in patients considered "non-blockers."[279]

Evidence Review Table 3. What are the best practices for preventing UTI associated with obstructed urinary catheters?

3.1.a. Further research is needed on the benefit of irrigating the catheter with acidifying solutions or use of oral urease inhibitors in long-term catheterized patients who have frequent catheter obstruction. **(No recommendation/unresolved issue)**

3.2.a. Silicone might be preferable to other materials to reduce the risk of encrustation in long-term catheterized patients who have frequent obstruction. **(Category II)**

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Management of Multidrug-Resistant Organisms In Healthcare Settings, 2006

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I. Introduction

Multidrug-resistant organisms(MDROs), including methicillin-resistant *Staphylococcus aureus* (MRSA), vancomycin-resistant enterococci (VRE) and certain gram-negative bacilli (GNB) have important infection control implications that either have not been addressed or received only limited consideration in previous isolation guidelines. Increasing experience with these organisms is improving understanding of the routes of transmission and effective preventive measures. Although transmission of MDROs is most frequently documented in acute care facilities, all healthcare settings are affected by the emergence and transmission of antimicrobial-resistant microbes. The severity and extent of disease caused by these pathogens varies by the population(s) affected and by the institution(s) in which they are found. Institutions, in turn, vary widely in physical and functional characteristics, ranging from long-term care facilities (LTCF) to specialty units (e.g., intensive care units [ICU], burn units, neonatal ICUs [NICUs]) in tertiary care facilities. Because of this, the approaches to prevention and control of these pathogens need to be tailored to the specific needs of each population and individual institution. The prevention and control of MDROs is a national priority - one that requires that all healthcare facilities and agencies assume responsibility(1) (2). The following discussion and recommendations are provided to guide the implementation of strategies and practices to prevent the transmission of MRSA, VRE, and other MDROs. The administration of healthcare organizations and institutions should ensure that appropriate strategies are fully implemented, regularly evaluated for effectiveness, and adjusted such that there is a consistent decrease in the incidence of targeted MDROs. Successful prevention and control of MDROs requires administrative and scientific leadership and a financial and human resource commitment(3-5). Resources must be made available for infection prevention and control, including expert consultation, laboratory support, adherence monitoring, and data analysis. Infection prevention and control professionals have found that healthcare personnel (HCP) are more receptive and adherent to the recommended control measures when organizational leaders participate in efforts to reduce MDRO transmission(3).

II. Background

MDRO definition*. For* epidemiologic purposes, MDROs are defined as microorganisms, predominantly bacteria, that are resistant to one or more classes of antimicrobial agents (1). Although the names of certain MDROs describe resistance to only one agent (e.g., MRSA, VRE), these pathogens are frequently resistant to most available antimicrobial agents . These highly resistant organisms deserve special attention in healthcare facilities (2). In addition to MRSA and VRE, certain GNB, including those producing extended spectrum beta-lactamases (ESBLs) and others that are resistant to multiple classes of antimicrobial agents, are of particular concern.[1] In addition to *Escherichia coli* and *Klebsiella pneumoniae*, these include strains of *Acinetobacter baumannii* resistant to all antimicrobial agents, or all except imipenem,(6-12), and organisms such as *Stenotrophomonas maltophilia* (12-14), *Burkholderia cepacia (15, 16)*, and *Ralstonia pickettii(17)* that are intrinsically resistant to the broadest-spectrum antimicrobial agents. In some residential settings (e.g., LTCFs), it is important to control multidrug-resistant *S. pneumoniae* (MDRSP) that are resistant to penicillin and other broad-spectrum agents such as macrolides and fluroquinolones (18, 19). Strains of *S. aureus* that have intermediate susceptibility or are resistant to vancomycin (i.e., vancomycin-intermediate *S. aureus* [VISA], vancomycin-resistant *S. aureus* [VRSA]) (20-30) have affected specific populations, such as hemodialysis patients.

Clinical importance of MDROs*.* In most instances, MDRO infections have clinical manifestations that are similar to infections caused by susceptible pathogens. However, options for treating patients with these infections are often extremely limited. For example, until recently, only vancomycin provided effective therapy for potentially life-threatening MRSA infections and during the 1990's there were virtually no antimicrobial agents to treat infections caused by VRE. Although antimicrobials are now available for treatment of MRSA and VRE infections, resistance to each new agent has already emerged in clinical

1 Multidrug-resistant strains of *M. tuberculosis* are not addressed in this document because of the markedly different patterns of transmission and spread of the pathogen and the very different control interventions that are needed for prevention of *M. tuberculosis* infection. Current recommendations for prevention and control of tuberculosis can be found at: http://www.cdc.gov/mmwr/pdf/rr/rr5417.pdf .

isolates(31-37). Similarly, therapeutic options are limited for ESBL-producing isolates of gram-negative bacilli, strains of *A. baumannii* resistant to all antimicrobial agents except imipenem(8-11, 38) and intrinsically resistant *Stenotrophomonas* sp.(12-14, 39). These limitations may influence antibiotic usage patterns in ways that suppress normal flora and create a favorable environment for development of colonization when exposed to potential MDR pathogens (i.e., selective advantage)(40).

Increased lengths of stay, costs, and mortality also have been associated with MDROs (41-46). Two studies documented increased mortality, hospital lengths of stay, and hospital charges associated with multidrug-resistant gram-negative bacilli (MDR-GNBs), including an NICU outbreak of ESBL-producing *Klebsiella pneumoniae* (47) and the emergence of third-generation cephalosporin resistance in *Enterobacter* spp. in hospitalized adults (48). Vancomycin resistance has been reported to be an independent predictor of death from enterococcal bacteremia(44, 49-53). Furthermore, VRE was associated with increased mortality, length of hospital stay, admission to the ICU, surgical procedures, and costs when VRE patients were compared with a matched hospital population (54).

However, MRSA may behave differently from other MDROs. When patients with MRSA have been compared to patients with methicillin-susceptible *S. aureus* (MSSA), MRSA-colonized patients more frequently develop symptomatic infections(55, 56). Furthermore, higher case fatality rates have been observed for certain MRSA infections, including bacteremia(57-62), poststernotomy mediastinitis(63), and surgical site infections(64). These outcomes may be a result of delays in the administration of vancomycin, the relative decrease in the bactericidal activity of vancomycin(65), or persistent bacteremia associated with intrinsic characteristics of certain MRSA strains (66). Mortality may be increased further by *S. aureus* with reduced vancomycin susceptibility (VISA) (26, 67). Also some studies have reported an association between MRSA infections and increased length of stay, and healthcare costs(46, 61, 62), while others have not(64). Finally, some hospitals have observed an increase in the overall occurrence of staphylococcal infections following the introduction of MRSA into a hospital or special-care unit(68, 69).

III. Epidemiology of MDROs

***Trends**:* Prevalence of MDROs varies temporally, geographically, and by healthcare setting(70, 71). For example, VRE emerged in the eastern United States in the early 1990s, but did not appear in the western United States until several years later, and MDRSP varies in prevalence by state(72). The type and level of care also influence the prevalence of MDROs. ICUs, especially those at tertiary care facilities, may have a higher prevalence of MDRO infections than do non-ICU settings (73, 74). Antimicrobial resistance rates are also strongly correlated with hospital size, tertiary-level care, and facility type (e.g., LTCF)(75, 76). The frequency of clinical infection caused by these pathogens is low in LTCFs(77, 78). Nonetheless, MDRO infections in LTCFs can cause serious disease and mortality, and colonized or infected LTCF residents may serve as reservoirs and vehicles for MDRO introduction into acute care facilities (78-88). Another example of population differences in prevalence of target MDROs is in the pediatric population. Point prevalence surveys conducted by the Pediatric Prevention Network (PPN) in eight U.S. PICUs and 7 U.S. NICUs in 2000 found $\leq$ 4% of patients were colonized with MRSA or VRE compared with 10-24% were colonized with ceftazidime- or aminoglycoside-resistant gram-negative bacilli; < 3% were colonized with ESBL-producing gram negative bacilli. Despite some evidence that MDRO burden is greatest in adult hospital patients, MDRO require similar control efforts in pediatric populations as well(89).

During the last several decades, the prevalence of MDROs in U.S. hospitals and medical centers has increased steadily(90, 91). MRSA was first isolated in the United States in 1968. By the early 1990s, MRSA accounted for 20%-25% of *Staphylococcus aureus* isolates from hospitalized patients(92). In 1999, MRSA accounted for >50% of *S. aureus* isolates from patients in ICUs in the National Nosocomial Infection Surveillance (NNIS) system; in 2003, 59.5% of *S. aureus* isolates in NNIS ICUs were MRSA (93). A similar rise in prevalence has occurred with VRE (94). From 1990 to 1997, the prevalence of VRE in enterococcal isolates from hospitalized patients increased from <1% to approximately 15% (95). VRE accounted for almost 25% of enterococcus isolates in NNIS ICUs in 1999 (94), and 28.5% in 2003 (93).

GNB resistant to ESBLs, fluoroquinolones, carbapenems, and aminoglycosides also have increased in prevalence. For example, in 1997, the SENTRY Antimicrobial Surveillance Program found that among *K. pneumoniae* strains isolated in the United States, resistance rates to ceftazidime and other third-generation cephalosporins were 6.6%, 9.7%, 5.4%, and 3.6% for bloodstream, pneumonia, wound, and urinary tract infections, respectively (95) In 2003, 20.6% of all *K. pneumoniae* isolates from NNIS ICUs were resistant to these drugs ((93)). Similarly, between 1999 and 2003, *Pseudomonas aeruginosa* resistance to fluoroquinolone antibiotics increased from 23% to 29.5% in NNIS ICUs(74). Also, a 3-month survey of 15 Brooklyn hospitals in 1999 found that 53% of *A. baumannii* strains exhibited resistance to carbapenems and 24% of *P. aeruginosa* strains were resistant to imipenem (10). During 1994-2000, a national review of ICU patients in 43 states found that the overall susceptibility to ciprofloxacin decreased from 86% to 76% and was temporally associated with increased use of fluoroquinolones in the United States (96).

Lastly, an analysis of temporal trends of antimicrobial resistance in non-ICU patients in 23 U.S. hospitals during 1996-1997 and 1998-1999 (97) found significant increases in the prevalence of resistant isolates including MRSA, ciprofloxacin-resistant *P. aeruginosa*, and ciprofloxacin- or ofloxacin-resistant *E. coli.* Several factors may have contributed to these increases including: selective pressure exerted by exposure to antimicrobial agents, particularly fluoroquinolones, outside of the ICU and/or in the community(7, 96, 98); increasing rates of community-associated MRSA colonization and infection(99, 100); inadequate adherence to infection control practices; or a combination of these factors.

Important concepts in transmission. Once MDROs are introduced into a healthcare setting, transmission and persistence of the resistant strain is determined by the availability of vulnerable patients, selective pressure exerted by antimicrobial use, increased potential for transmission from larger numbers of colonized or infected patients (“colonization pressure”)(101, 102); and the impact of implementation and adherence to prevention efforts. Patients vulnerable to colonization and infection include those with severe disease, especially those with compromised host defenses from underlying medical conditions; recent surgery; or indwelling medical devices (e.g., urinary catheters or endotracheal

tubes(103, 104)). Hospitalized patients, especially ICU patients, tend to have more risk factors than non-hospitalized patients do, and have the highest infection rates. For example, the risk that an ICU patient will acquire VRE increases significantly once the proportion of ICU patients colonized with VRE exceeds 50%(101) or the number days of exposure to a VRE-patient exceeds 15 days(105). A similar effect of colonization pressure has been demonstrated for MRSA in a medical ICU(102). Increasing numbers of infections with MDROs also have been reported in non-ICU areas of hospitals(97).

There is ample epidemiologic evidence to suggest that MDROs are carried from one person to another via the hands of HCP(106-109). Hands are easily contaminated during the process of care-giving or from contact with environmental surfaces in close proximity to the patient(110-113). The latter is especially important when patients have diarrhea and the reservoir of the MDRO is the gastrointestinal tract(114-117). Without adherence to published recommendations for hand hygiene and glove use(111) HCP are more likely to transmit MDROs to patients. Thus, strategies to increase and monitor adherence are important components of MDRO control programs(106, 118).

Opportunities for transmission of MDROs beyond the acute care hospital results from patients receiving care at multiple healthcare facilities and moving between acute-care, ambulatory and/or chronic care, and LTC environments. System-wide surveillance at LDS Hospital in Salt Lake City, Utah, monitored patients identified as being infected or colonized with MRSA or VRE, and found that those patients subsequently received inpatient or outpatient care at as many as 62 different healthcare facilities in that system during a 5-year span(119).

Role of colonized HCP in MDRO transmission. Rarely, HCP may introduce an MDRO into a patient care unit(120-123). Occasionally, HCP can become persistently colonized with an MDRO, but these HCP have a limited role in transmission, unless other factors are present. Additional factors that can facilitate transmission, include chronic sinusitis(120), upper respiratory infection(123), and dermatitis(124).

Implications of community-associated MRSA (CA-MRSA). The emergence of new epidemic strains of MRSA in the community, among patients without established MRSA risk factors, may present new challenges to MRSA control in healthcare settings(125-128). Historically, genetic analyses of MRSA isolated from patients in hospitals worldwide revealed that a relatively small number of MRSA strains have unique qualities that facilitate their transmission from patient to patient within healthcare facilities over wide geographic areas, explaining the dramatic increases in HAIs caused by MRSA in the 1980s and early 1990s(129). To date, most MRSA strains isolated from patients with CA-MRSA infections have been microbiologically distinct from those endemic in healthcare settings, suggesting that some of these strains may have arisin *de novo* in the community via acquisition of methicillin resistance genes by established methicillin-susceptible *S. aureus* (MSSA) strains(130-132). Two pulsed-field types, termed USA300 and USA400 according to a typing scheme established at CDC, have accounted for the majority of CA-MRSA infections characterized in the United States, whereas pulsed-field types USA100 and USA200 are the predominant genotypes endemic in healthcare settings(133).

USA300 and USA400 genotypes almost always carry type IV of the staphylococcal chromosomal cassette (SCC) *mec*, the mobile genetic element that carries the *mec*A methicillin-resistance gene (133, 134). This genetic cassette is smaller than types I through III, the types typically found in healthcare associated MRSA strains, and is hypothesized to be more easily transferable between *S. aureus* strains.

CA-MRSA infection presents most commonly as relatively minor skin and soft tissue infections, but severe invasive disease, including necrotizing pneumonia, necrotizing fasciitis, severe osteomyelitis, and a sepsis syndrome with increased mortality have also been described in children and adults(134-136).

Transmission within hospitals of MRSA strains first described in the community (e.g. USA300 and USA400) are being reported with increasing frequency(137-140). Changing resistance patterns of MRSA in ICUs in the NNIS system from 1992 to 2003 provide additional evidence that the new epidemic MRSA strains are becoming established

healthcare-associated as well as community pathogens(90). Infections with these strains have most commonly presented as skin disease in community settings. However, intrinsic virulence characteristics of the organisms can result in clinical manifestations similar to or potentially more severe than traditional healthcare-associated MRSA infections among hospitalized patients. The prevalence of MRSA colonization and infection in the surrounding community may therefore affect the selection of strategies for MRSA control in healthcare settings.

IV. MDRO Prevention and Control

Prevention of Infections. Preventing infections will reduce the burden of MDROs in healthcare settings. Prevention of antimicrobial resistance depends on appropriate clinical practices that should be incorporated into all routine patient care. These include optimal management of vascular and urinary catheters, prevention of lower respiratory tract infection in intubated patients, accurate diagnosis of infectious etiologies, and judicious antimicrobial selection and utilization. Guidance for these preventive practices include the Campaign to Reduce Antimicrobial Resistance in Healthcare Settings (www.cdc.gov/drugresistance/healthcare/default.htm), a multifaceted, evidence-based approach with four parallel strategies: infection prevention; accurate and prompt diagnosis and treatment; prudent use of antimicrobials; and prevention of transmission. Campaign materials are available for acute care hospitals, surgical settings, dialysis units, LTCFs and pediatric acute care units.

To reduce rates of central-venous-line associated bloodstream infections(CVL-BSIs) and ventilator-associated pneumonia (VAP), a group of bundled evidence-based clinical practices have been implemented in many U.S. healthcare facilities(118, 141-144). One report demonstrated a sustained effect on the reduction in CVL-BSI rates with this approach(145). Although the specific effect on MDRO infection and colonization rates have not been reported, it is logical that decreasing these and other healthcare-associated infections will in turn reduce antimicrobial use and decrease opportunities for emergence and transmission of MDROs.

Prevention and Control of MDRO transmission

Overview of the MDRO control literature. Successful control of MDROs has been documented in the United States and abroad using a variety of combined interventions. These include improvements in hand hygiene, use of Contact Precautions until patients are culture-negative for a target MDRO, active surveillance cultures (ASC), education, enhanced environmental cleaning, and improvements in communication about patients with MDROs within and between healthcare facilities.

Representative studies include:

- Reduced rates of MRSA transmission in The Netherlands, Belgium, Denmark, and other Scandinavian countries after the implementation of aggressive and sustained infection control interventions (i.e., ASC; preemptive use of Contact Precautions upon admission until proven culture negative; and, in some instances, closure of units to new admissions). MRSA generally accounts for a very small proportion of *S. aureus* clinical isolates in these countries(146-150).
- Reduced rates of VRE transmission in healthcare facilities in the three-state Siouxland region (Iowa, Nebraska, and South Dakota) following formation of a coalition and development of an effective region-wide infection control intervention that included ASC and isolation of infected patients. The overall prevalence rate of VRE in the 30 participating facilities decreased from 2.2% in 1997 to 0.5% in 1999(151).
- Eradication of endemic MRSA infections from two NICUs. The first NICU included implementation of ASC, Contact Precautions, use of triple dye on the umbilical cord, and systems changes to improve surveillance and adherence to recommended practices and to reduce overcrowding(152). The second NICU used ASC and Contact Precautions; surgical masks were included in the barriers used for Contact Precautions(153).
- Control of an outbreak and eventual eradication of VRE from a burn unit over a 13-month period with implementation of aggressive culturing, environmental cleaning, and barrier isolation(154).
- Control of an outbreak of VRE in a NICU over a 3-year period with implementation of ASC, other infection control measures such as use of a waterless hand disinfectant, and mandatory in-service education(155).

- Eradication of MDR-strains of *A. baumannii* from a burn unit over a 16-month period with implementation of strategies to improve adherence to hand hygiene, isolation, environmental cleaning, and temporary unit closure(38).
- In addition, more than 100 reports published during 1982-2005 support the efficacy of combinations of various control interventions to reduce the burden of MRSA, VRE, and MDR-GNBs (Tables 1 and 2). Case-rate reduction or pathogen eradication was reported in a majority of studies.
- VRE was eradicated in seven special-care units(154, 156-160), two hospitals(161, 162), and one LTCF(163).
- MRSA was eradicated from nine special-care units(89, 152, 153, 164-169), two hospitals(170), one LTCF(167), and one Finnish district(171). Furthermore, four MRSA reports described continuing success in sustaining low endemic MDRO rates for over 5 years(68, 166, 172, 173).
- An MDR-GNB was eradicated from 13 special-care units(8, 9, 38, 174-180) and two hospitals (11, 181).

These success stories testify to the importance of having dedicated and knowledgeable teams of healthcare professionals who are willing to persist for years, if necessary, to control MDROs. Eradication and control of MDROs, such as those reported, frequently required periodic reassessment and the addition of new and more stringent interventions over time (tiered strategy). For example, interventions were added in a stepwise fashion during a 3-year effort that eventually eradicated MRSA from an NICU(152). A series of interventions was adopted throughout the course of a year to eradicate VRE from a burn unit(154). Similarly, eradication of carbapenem-resistant strains of *A. baumannii* from a hospital required multiple and progressively more intense interventions over several years(11).

Nearly all studies reporting successful MDRO control employed a median of 7 to 8 different interventions concurrently or sequentially (Table 1). These figures may underestimate the actual number of control measures used, because authors of these reports may have considered their earliest efforts routine (e.g., added emphasis on handwashing), and did not include them as interventions, and some ”single measures” are, in fact, a complex

combination of several interventions. The use of multiple concurrent control measures in these reports underscores the need for a comprehensive approach for controlling MDROs.

Several factors affect the ability to generalize the results of the various studies reviewed, including differences in definition, study design, endpoints and variables measured, and period of follow-up. Two-thirds of the reports cited in Tables 1 and 2 involved perceived outbreaks, and one-third described efforts to reduce endemic transmission. Few reports described preemptive efforts or prospective studies to control MDROs before they had reached high levels within a unit or facility.

With these and other factors, it has not been possible to determine the effectiveness of individual interventions, or a specific combination of interventions, that would be appropriate for all healthcare facilities to implement in order to control their target MDROs. Randomized controlled trials are necessary to acquire this level of evidence. An NIH-sponsored, randomized controlled trial on the prevention of MRSA and VRE transmission in adult ICUs is ongoing and may provide further insight into optimal control measures (http://clinicaltrials.gov/ct/show/NCT00100386?order=1). This trial compares the use of education (to improve adherence to hand hygiene) and Standard Precautions to the use of ASC and Contact Precautions.

Control Interventions. The various types of interventions used to control or eradicate MDROs may be grouped into seven categories. These include administrative support, judicious use of antimicrobials, surveillance (routine and enhanced), Standard and Contact Precautions, environmental measures, education and decolonization. These interventions provide the basis for the recommendations for control of MDROs in healthcare settings that follow this review and as summarized in Table 3. In the studies reviewed, these interventions were applied in various combinations and degrees of intensity, with differences in outcome.

1. ***Administrative support.*** In several reports, administrative support and involvement were important for the successful control of the target MDRO(3, 152, 182-185), and authorities in infection control have strongly recommended such support(2, 106, 107,

186). There are several examples of MDRO control interventions that require administrative commitment of fiscal and human resources. One is the use of ASC(8, 38, 68, 107, 114, 151, 152, 167, 168, 183, 184, 187-192). Other interventions that require administrative support include: 1) implementing system changes to ensure prompt and effective communications e.g., computer alerts to identify patients previously known to be colonized/infected with MDROs(184, 189, 193, 194); 2), providing the necessary number and appropriate placement of hand washing sinks and alcohol-containing hand rub dispensers in the facility(106, 195); 3) maintaining staffing levels appropriate to the intensity of care required(152, 196-202); and 4) enforcing adherence to recommended infection control practices (e.g., hand hygiene, Standard and Contact Precautions) for MDRO control. Other measures that have been associated with a positive impact on prevention efforts, that require administrative support, are direct observation with feedback to HCP on adherence to recommended precautions and keeping HCP informed about changes in transmission rates(3, 152, 182, 203-205). A “How-to guide” for implementing change in ICUs, including analysis of structure, process, and outcomes when designing interventions, can assist in identification of needed administrative interventions(195). Lastly, participation in existing, or the creation of new, city-wide, state-wide, regional or national coalitions, to combat emerging or growing MDRO problems is an effective strategy that requires administrative support(146, 151, 167, 188, 206, 207).

2. ***Education.*** Facility-wide, unit-targeted, and informal, educational interventions were included in several successful studies(3, 189, 193, 208-211). The focus of the interventions was to encourage a behavior change through improved understanding of the problem MDRO that the facility was trying to control. Whether the desired change involved hand hygiene, antimicrobial prescribing patterns, or other outcomes, enhancing understanding and creating a culture that supported and promoted the desired behavior, were viewed as essential to the success of the intervention. Educational campaigns to enhance adherence to hand hygiene practices in conjunction with other control measures have been associated temporally with decreases in MDRO transmission in various healthcare settings(3, 106, 163).

3. ***Judicious use of antimicrobial agents.*** While a comprehensive review of antimicrobial stewardship is beyond the scope of this guideline, recommendations for control of MDROs must include attention to judicious antimicrobial use. A temporal association between formulary changes and decreased occurrence of a target MDRO was found in several studies, especially in those that focused on MDR-GNBs(98, 177, 209, 212-218). Occurrence of C. difficile-associated disease has also been associated with changes in antimicrobial use(219). Although some MRSA and VRE control efforts have attempted to limit antimicrobial use, the relative importance of this measure for controlling these MDROs remains unclear(193, 220). Limiting antimicrobial use alone may fail to control resistance due to a combination of factors; including 1) the relative effect of antimicrobials on providing initial selective pressure, compared to perpetuating resistance once it has emerged; 2) inadequate limits on usage; or 3) insufficient time to observe the impact of this intervention. With the intent of addressing #2 and #3 above in the study design, one study demonstrated a decrease in the prevalence of VRE associated with a formulary switch from ticarcillin-clavulanate to piperacillin-tazobactam(221).

   The CDC Campaign to Prevent Antimicrobial Resistance that was launched in 2002 provides evidence-based principles for judicious use of antimicrobials and tools for implementation(222) www.cdc.gov/drugresistance/healthcare. This effort targets all healthcare settings and focuses on effective antimicrobial treatment of infections, use of narrow spectrum agents, treatment of infections and not contaminants, avoiding excessive duration of therapy, and restricting use of broad-spectrum or more potent antimicrobials to treatment of serious infections when the pathogen is not known or when other effective agents are unavailable. Achieving these objectives would likely diminish the selective pressure that favors proliferation of MDROs. Strategies for influencing antimicrobial prescribing patterns within healthcare facilities include education; formulary restriction; prior-approval programs, including pre-approved indications; automatic stop orders; academic interventions to counteract pharmaceutical influences on prescribing patterns; antimicrobial cycling(223-226);

computer-assisted management programs(227-229); and active efforts to remove redundant antimicrobial combinations(230). A systematic review of controlled studies identified several successful practices. These include social marketing (i.e. consumer education), practice guidelines, authorization systems, formulary restriction, mandatory consultation, and peer review and feedback. It further suggested that online systems that provide clinical information, structured order entry, and decision support are promising strategies(231). These changes are best accomplished through an organizational, multidisciplinary, antimicrobial management program(232).

4. ***MDRO surveillance.*** Surveillance is a critically important component of any MDRO control program, allowing detection of newly emerging pathogens, monitoring epidemiologic trends, and measuring the effectiveness of interventions. Multiple MDRO surveillance strategies have been employed, ranging from surveillance of clinical microbiology laboratory results obtained as part of routine clinical care, to use of ASC to detect asymptomatic colonization.

Surveillance for MDROs isolated from routine clinical cultures.

Antibiograms. The simplest form of MDRO surveillance is monitoring of clinical microbiology isolates resulting from tests ordered as part of routine clinical care. This method is particularly useful to detect emergence of new MDROs not previously detected, either within an individual healthcare facility or community-wide. In addition, this information can be used to prepare facility- or unit-specific summary antimicrobial susceptibility reports that describe pathogen-specific prevalence of resistance among clinical isolates. Such reports may be useful to monitor for changes in known resistance patterns that might signal emergence or transmission of MDROs, and also to provide clinicians with information to guide antimicrobial prescribing practices(233-235).

MDRO Incidence Based on Clinical Culture Results. Some investigators have used clinical microbiology results to calculate measures of incidence of MDRO isolates in specific populations or patient care locations (e.g. new MDRO

isolates/1,000 patient days, new MDRO isolates per month)(205, 236, 237). Such measures may be useful for monitoring MDRO trends and assessing the impact of prevention programs, although they have limitations. Because they are based solely on positive culture results without accompanying clinical information, they do not distinguish colonization from infection, and may not fully demonstrate the burden of MDRO-associated disease. Furthermore, these measures do not precisely measure acquisition of MDRO colonization in a given populaton or location. Isolating an MDRO from a clinical culture obtained from a patient several days after admission to a given unit or facility does not establish that the patient acquired colonization in that unit. On the other hand, patients who acquire MDRO colonization may remain undetected by clinical cultures(107). Despite these limitations, incidence measures based on clinical culture results may be highly correlated with actual MDRO transmission rates derived from information using ASC, as demonstrated in a recent multicenter study(237). These results suggest that incidence measures based on clinical cultures alone might be useful surrogates for monitoring changes in MDRO transmission rates.

MDRO Infection Rates. Clinical cultures can also be used to identify targeted MDRO infections in certain patient populations or units(238, 239). This strategy requires investigation of clinical circumstances surrounding a positive culture to distinguish colonization from infection, but it can be particularly helpful in defining the clinical impact of MDROs within a facility.

Molecular typing of MDRO isolates. Many investigators have used molecular typing of selected isolates to confirm clonal transmission to enhance understanding of MDRO transmission and the effect of interventions within their facility(38, 68, 89, 92, 138, 152, 190, 193, 236, 240).

Surveillance for MDROs by Detecting Asymptomatic Colonization

Another form of MDRO surveillance is the use of active surveillance cultures (ASC) to identify patients who are colonized with a targeted MDRO(38, 107, 241). This

approach is based upon the observation that, for some MDROs, detection of colonization may be delayed or missed completely if culture results obtained in the course of routine clinical care are the primary means of identifying colonized patients(8, 38, 107, 114, 151, 153, 167, 168, 183, 184, 187, 189, 191-193, 242-244). Several authors report having used ASC when new pathogens emerge in order to define the epidemiology of the particular agent(22, 23, 107, 190). In addition, the authors of several reports have concluded that ASC, in combination with use of Contact Precautions for colonized patients, contributed directly to the decline or eradication of the target MDRO(38, 68, 107, 151, 153, 184, 217, 242). However, not all studies have reached the same conclusion. Poor control of MRSA despite use of ASC has been described(245). A recent study failed to identify cross-transmission of MRSA or MSSA in a MICU during a 10 week period when ASC were obtained, despite the fact that culture results were not reported to the staff(246). The investigators suggest that the degree of cohorting and adherence to Standard Precautions might have been the important determinants of transmission prevention, rather than the use of ASC and Contact Precautions for MRSA-colonized patients. The authors of a systematic review of the literature on the use of isolation measures to control healthcare-associated MRSA concluded that there is evidence that concerted efforts that include ASC and isolation can reduce MRSA even in endemic settings. However, the authors also noted that methodological weaknesses and inadequate reporting in published research make it difficult to rule out plausible alternative explanations for reductions in MRSA acquisition associated with these interventions, and therefore concluded that the precise contribution of active surveillance and isolation alone is difficult to assess(247).

Mathematical modeling studies have been used to estimate the impact of ASC use in control of MDROs. One such study evaluating interventions to decrease VRE transmission indicated that use of ASC (versus no cultures) could potentially decrease transmission 39% and that with pre-emptive isolation plus ASC, transmission could be decreased 65%(248). Another mathematical model examining the use of ASC and isolation for control of MRSA predicted that isolating colonized or

infected patients on the basis of clinical culture results is unlikely to be successful at controlling MRSA, whereas use of active surveillance and isolation can lead to successful control, even in settings where MRSA is highly endemic.(249) There is less literature on the use of ASC in controlling MDR-GNBs. Active surveillance cultures have been used as part of efforts to successful control of MDR-GNBs in outbreak settings. The experience with ASC as part of successful control efforts in endemic settings is mixed. One study reported successful reduction of extended-spectrum beta-lactamase –producing Enterobacteriaceae over a six year period using a multifaceted control program that included use of ASC(245). Other reports suggest that use of ASC is not necessary to control endemic MDR-GNBs.(250, 251).

More research is needed to determine the circumstances under which ASC are most beneficial(252), but their use should be considered in some settings, especially if other control measures have been ineffective. When use of ASC is incorporated into MDRO prevention programs, the following should be considered:

- The decision to use ASC as part of an infection prevention and control program requires additional support for successful implementation, including: 1) personnel to obtain the appropriate cultures, 2) microbiology laboratory personnel to process the cultures, 3) mechanism for communicating results to caregivers, 4) concurrent decisions about use of additional isolation measures triggered by a positive culture (e.g. Contact Precautions) and 5) mechanism for assuring adherence to the additional isolation measures.
- The populations targeted for ASC are not well defined and vary among published reports. Some investigators have chosen to target specific patient populations considered at high risk for MDRO colonization based on factors such as location (e.g. ICU with high MDRO rates), antibiotic exposure history, presence of underlying diseases, prolonged duration of stay, exposure to other MDRO-colonized patients, patients transferred from other facilities known to have a high prevalence of MDRO carriage, or having a history of recent hospital or nursing home stays(107, 151, 253). A more commonly employed strategy involves obtaining surveillance cultures from all patients admitted to units experiencing

high rates of colonization/infection with the MDROs of interest, unless they are already known to be MDRO carriers(153, 184, 242, 254). In an effort to better define target populations for active surveillance, investigators have attempted to create prediction rules to identify subpopulations of patients at high risk for colonization on hospital admission(255, 256). Decisions about which populations should be targeted for active surveillance should be made in the context of local determinations of the incidence and prevalence of MDRO colonization within the intervention facility as well as other facilities with whom patients are frequently exchanged(257).

- Optimal timing and interval of ASC are not well defined. In many reports, cultures were obtained at the time of admission to the hospital or intervention unit or at the time of transfer to or from designated units (e.g., ICU)(107). In addition, some hospitals have chosen to obtain cultures on a periodic basis [e.g., weekly(8, 153, 159) to detect silent transmission. Others have based follow-up cultures on the presence of certain risk factors for MDRO colonization, such as antibiotic exposure, exposure to other MDRO colonized patients, or prolonged duration of stay in a high risk unit(253).
- Methods for obtaining ASC must be carefully considered, and may vary depending upon the MDRO of interest.
    - MRSA: Studies suggest that cultures of the nares identify most patients with MRSA and perirectal and wound cultures can identify additional carriers(152, 258-261).
    - VRE: Stool, rectal, or perirectal swabs are generally considered a sensitive method for detection of VRE. While one study suggested that rectal swabs may identify only 60% of individuals harboring VRE, and may be affected by VRE stool density(262), this observation has not been reported elsewhere in the literature.
    - MDR-GNBs: Several methods for detection of MDR-GNBs have been employed, including use of peri-rectal or rectal swabs alone or in combination with oro-pharyngeal, endotracheal, inguinal, or wound cultures. The absence of standardized screening media for many gram-

negative bacilli can make the process of isolating a specific MDR-GNB a relatively labor-intensive process(38, 190, 241, 250).

- Rapid detection methods: Using conventional culture methods for active surveillance can result in a delay of 2-3 days before results are available. If the infection control precautions (e.g., Contact Precautions) are withheld until the results are available, the desired infection control measures could be delayed. If empiric precautions are used pending negative surveillance culture results, precautions may be unnecessarily implemented for many, if not most, patients. For this reason, investigators have sought methods for decreasing the time necessary to obtain a result from ASC. Commercially available media containing chromogenic enzyme substrates (CHROMagar MRSA(263, 264) has been shown to have high sensitivity and specificity for identification of MRSA and facilitate detection of MRSA colonies in screening cultures as early as 16 hours after inoculation. In addition, real-time PCR-based tests for rapid detection of MRSA directly from culture swabs (< 1-2 hours) are now commercially available(265-267), as well as PCR-based tests for detection of vanA and van B genes from rectal swabs(268). The impact of rapid testing on the effectiveness of active surveillance as a prevention strategy, however, has not been fully determined. Rapid identification of MRSA in one study was associated with a significant reduction in MRSA infections acquired in the medical ICU, but not the surgical ICU(265). A mathematical model characterizing MRSA transmission dynamics predicted that, in comparison to conventional culture methods, the use of rapid detection tests may decrease isolation needs in settings of low-endemicity and result in more rapid reduction in prevalence in highly-endemic settings(249).

- Some MDRO control reports described surveillance cultures of healthcare personnel during outbreaks, but colonized or infected healthcare personnel are rarely the source of ongoing transmission, and this strategy should be reserved for settings in which specific healthcare personnel have been epidemiologically implicated in the transmission of MDROs(38, 92, 152-154, 188).

5. ***Infection Control Precautions.*** Since 1996 CDC has recommended the use of Standard and Contact Precautions for MDROs "judged by an infection control program…to be of special clinical and epidemiologic significance." This recommendation was based on general consensus and was not necessarily evidence-based. No studies have directly compared the efficacy of Standard Precautions alone versus Standard Precautions and Contact Precautions, with or without ASC, for control of MDROs. Some reports mention the use of one or both sets of precautions as part of successful MDRO control efforts; however, the precautions were not the primary focus of the study intervention(164, 190, 205, 269-271). The NIH-sponsored study mentioned earlier (Section: *Overview of the MDRO control literature*) may provide some answers, http://clinicaltrials.gov/ct/show/NCT00100386?order=1).

   Standard Precautions have an essential role in preventing MDRO transmission, even in facilities that use Contact Precautions for patients with an identified MDRO. Colonization with MDROs is frequently undetected; even surveillance cultures may fail to identify colonized persons due to lack of sensitivity, laboratory deficiencies, or intermittent colonization due to antimicrobial therapy(262). Therefore, Standard Precautions must be used in order to prevent transmission from potentially colonized patients. Hand hygiene is an important component of Standard Precautions. The authors of the *Guideline for Hand Hygiene in Healthcare Settings(106)* cited nine studies that demonstrated a temporal relationship between improved adherence to recommended hand hygiene practices and control of MDROs. It is noteworthy that in one report the frequency of hand hygiene did not improve with use of Contact Precautions but did improve when gloves were used (per Standard Precautions) for contact with MDRO patients(272).

   MDRO control efforts frequently involved changes in isolation practices, especially during outbreaks. In the majority of reports, Contact Precautions were implemented for all patients found to be colonized or infected with the target MDRO (See Table 2).

Some facilities also preemptively used Contact Precautions, in conjunction with ASC, for all new admissions or for all patients admitted to a specific unit, until a negative screening culture for the target MDRO was reported(30, 184, 273).

Contact Precautions are intended to prevent transmission of infectious agents, including epidemiologically important microorganisms, which are transmitted by direct or indirect contact with the patient or the patient's environment. A single-patient room is preferred for patients who require Contact Precautions. When a single-patient room is not available, consultation with infection control is necessary to assess the various risks associated with other patient placement options (e.g., cohorting, keeping the patient with an existing roommate). HCP caring for patients on Contact Precautions should wear a gown and gloves for all interactions that may involve contact with the patient or potentially contaminated areas in the patient's environment. Donning gown and gloves upon room entry and discarding before exiting the patient room is done to contain pathogens, especially those that have been implicated in transmission through environmental contamination (e.g., VRE, *C. difficile,* noroviruses and other intestinal tract agents; RSV)(109, 111, 274-277).

Cohorting and other MDRO control strategies. In several reports, cohorting of patients(152, 153, 167, 183, 184, 188, 189, 217, 242), cohorting of staff(184, 217, 242, 278), use of designated beds or units(183, 184), and even unit closure(38, 146, 159, 161, 279, 280) were necessary to control transmission. Some authors indicated that implementation of the latter two strategies were the turning points in their control efforts; however, these measures usually followed many other actions to prevent transmission. In one, two-center study, moving MRSA-positive patients into single rooms or cohorting these patients in designated bays failed to reduce transmission in ICUs. However, in this study adherence to recommendations for hand hygiene between patient contacts was only 21%(281). Other published studies, including one commissioned by the American Institute of Architects and the Facility Guidelines Institute (www.aia.org/aah_gd_hospcons), have documented a beneficial relationship between private rooms and reduction in risk of acquiring MDROs(282). Additional

studies are needed to define the specific contribution of using single-patient rooms and/or cohorting on preventing transmission of MDROs.

Duration of Contact Precautions. The necessary duration of Contact Precautions for patients treated for infection with an MDRO, but who may continue to be colonized with the organism at one or more body sites, remains an unresolved issue. Patients may remain colonized with MDROs for prolonged periods; shedding of these organisms may be intermittent, and surveillance cultures may fail to detect their presence(84, 250, 283). The 1995 HICPAC guideline for preventing the transmission of VRE suggested three negative stool/perianal cultures obtained at weekly intervals as a criterion for discontinuation of Contact Precautions(274). One study found these criteria generally reliable(284). However, this and other studies have noted a recurrence of VRE positive cultures in persons who subsequently receive antimicrobial therapy and persistent or intermittent carriage of VRE for more than 1 year has been reported(284-286). Similarly, colonization with MRSA can be prolonged(287, 288). Studies demonstrating initial clearance of MRSA following decolonization therapy have reported a high frequency of subsequent carriage(289, 290). There is a paucity of information in the literature on when to discontinue Contact Precautions for patients colonized with a MDR-GNB, possibly because infection and colonization with these MDROs are often associated with outbreaks. Despite the uncertainty about when to discontinue Contact Precautions, the studies offer some guidance. In the context of an outbreak, prudence would dictate that Contact Precautions be used indefinitely for all previously infected and known colonized patients. Likewise, if ASC are used to detect and isolate patients colonized with MRSA or VRE, and there is no decolonization of these patients, it is logical to assume that Contact Precautions would be used for the duration of stay in the setting where they were first implemented. In general, it seems reasonable to discontinue Contact Precautions when three or more surveillance cultures for the target MDRO are repeatedly negative over the course of a week or two in a patient who has not received antimicrobial therapy for several weeks, especially in the absence of a

draining wound, profuse respiratory secretions, or evidence implicating the specific patient in ongoing transmission of the MDRO within the facility.

Barriers used for contact with patients infected or colonized with MDROs. Three studies evaluated the use of gloves with or without gowns for all patient contacts to prevent VRE acquisition in ICU settings(30, 105, 273). Two of the studies showed that use of both gloves and gowns reduced VRE transmission(30, 105) while the third showed no difference in transmission based on the barriers used(273). One study in a LTCF compared the use of gloves only, with gloves plus contact isolation, for patients with four MDROs, including VRE and MRSA, and found no difference(86). However, patients on contact isolation were more likely to acquire MDR-*K. pneumoniae* strains that were prevalent in the facility; reasons for this were not specifically known. In addition to differences in outcome, differing methodologies make comparisons difficult. Specifically, HCP adherence to the recommended protocol, the influence of added precautions on the number of HCP-patient interactions, and colonization pressure were not consistently assessed.

Impact of Contact Precautions on patient care and well-being. There are limited data regarding the impact of Contact Precautions on patients. Two studies found that HCP, including attending physicians, were half as likely to enter the rooms of(291), or examine(292), patients on Contact Precautions. Other investigators have reported similar observations on surgical wards(293). Two studies reported that patients in private rooms and on barrier precautions for an MDRO had increased anxiety and depression scores(294, 295). Another study found that patients placed on Contact Precautions for MRSA had significantly more preventable adverse events, expressed greater dissatisfaction with their treatment, and had less documented care than control patients who were not in isolation(296). Therefore, when patients are placed on Contact Precautions, efforts must be made by the healthcare team to counteract these potential adverse effects.

6. ***Environmental measures.*** The potential role of environmental reservoirs, such as surfaces and medical equipment, in the transmission of VRE and other MDROs has been the subject of several reports(109-111, 297, 298). While environmental cultures are not routinely recommended(299), environmental cultures were used in several studies to document contamination, and led to interventions that included the use of dedicated noncritical medical equipment(217, 300), assignment of dedicated cleaning personnel to the affected patient care unit(154), and increased cleaning and disinfection of frequently-touched surfaces (e.g., bedrails, charts, bedside commodes, doorknobs). A common reason given for finding environmental contamination with an MDRO was the lack of adherence to facility procedures for cleaning and disinfection. In an educational and observational intervention, which targeted a defined group of housekeeping personnel, there was a persistent decrease in the acquisition of VRE in a medical ICU(301). Therefore, monitoring for adherence to recommended environmental cleaning practices is an important determinant for success in controlling transmission of MDROs and other pathogens in the environment(274, 302).

   In the MDRO reports reviewed, enhanced environmental cleaning was frequently undertaken when there was evidence of environmental contamination and ongoing transmission. Rarely, control of the target MDRO required vacating a patient care unit for complete environmental cleaning and assessment(175, 279).

7. ***Decolonization.*** Decolonization entails treatment of persons colonized with a specific MDRO, usually MRSA, to eradicate carriage of that organism. Although some investigators have attempted to decolonize patients harboring VRE(220), few have achieved success. However, decolonization of persons carrying MRSA in their nares has proved possible with several regimens that include topical mupirocin alone or in combination with orally administered antibiotics (e.g., rifampin in combination with trimethoprim- sulfamethoxazole or ciprofloxacin) plus the use of an antimicrobial soap for bathing(303). In one report, a 3-day regimen of baths with povidone-iodine and nasal therapy with mupirocin resulted in eradication of nasal MRSA

colonization(304). These and other methods of MRSA decolonization have been thoroughly reviewed.(303, 305-307).

Decolonization regimens are not sufficiently effective to warrant routine use. Therefore, most healthcare facilities have limited the use of decolonization to MRSA outbreaks, or other high prevalence situations, especially those affecting special-care units. Several factors limit the utility of this control measure on a widespread basis: 1) identification of candidates for decolonization requires surveillance cultures; 2) candidates receiving decolonization treatment must receive follow-up cultures to ensure eradication; and 3) recolonization with the same strain, initial colonization with a mupirocin-resistant strain, and emergence of resistance to mupirocin during treatment can occur(289, 303, 308-310). HCP implicated in transmission of MRSA are candidates for decolonization and should be treated and culture negative before returning to direct patient care. In contrast, HCP who are colonized with MRSA, but are asymptomatic, and have not been linked epidemiologically to transmission, do not require decolonization.

IV. Discussion

This review demonstrates the depth of published science on the prevention and control of MDROs. Using a combination of interventions, MDROs in endemic, outbreak, and non-endemic settings have been brought under control. However, despite the volume of literature, an appropriate set of evidence-based control measures that can be universally applied in all healthcare settings has not been definitively established. This is due in part to differences in study methodology and outcome measures, including an absence of randomized, controlled trials comparing one MDRO control measure or strategy with another. Additionally, the data are largely descriptive and quasi-experimental in design(311). Few reports described preemptive efforts or prospective studies to control MDROs before they had reached high levels within a unit or facility. Furthermore, small hospitals and LTCFs are infrequently represented in the literature.
A number of questions remain and are discussed below.

Impact on other MDROS from interventions targeted to one MDRO Only one report described control efforts directed at more than one MDRO, i.e., MDR-GNB and MRSA(312). Several reports have shown either decreases or increases in other pathogens with efforts to control one MDRO. For example, two reports on VRE control efforts demonstrated an increase in MRSA following the prioritization of VRE patients to private rooms and cohort beds(161). Similarly an outbreak of *Serratia marcescens* was temporally associated with a concurrent, but unrelated, outbreak of MRSA in an NICU(313). In contrast, Wright and colleagues reported a decrease in MRSA and VRE acquisition in an ICU during and after their successful effort to eradicate an MDR-strain of *A. baumannii* from the unit(210).

Colonization with multiple MDROs appears to be common(314, 315). One study found that nearly 50% of residents in a skilled-care unit in a LTCF were colonized with a target MDRO and that 26% were co-colonized with >1 MDRO; a detailed analysis showed that risk factors for colonization varied by pathogen(316). One review of the literature(317) reported that patient risk factors associated with colonization with MRSA, VRE, MDR-GNB, *C. difficile* and *Candida sp* were the same. This review concluded that control programs that focus on only one organism or one antimicrobial drug are unlikely to succeed because vulnerable patients will continue to serve as a magnet for other MDROs.

Costs. Several authors have provided evidence for the cost-effectiveness of approaches that use ASC(153, 191, 253, 318, 319). However, the supportive evidence often relied on assumptions, projections, and estimated attributable costs of MDRO infections. Similar limitations apply to a study suggesting that gown use yields a cost benefit in controlling transmission of VRE in ICUs(320). To date, no studies have directly compared the benefits and costs associated with different MDRO control strategies.

Feasibility. The subject of feasibility, as it applies to the extrapolation of results to other healthcare settings, has not been addressed. For example, smaller hospitals and LTCFs may lack the on-site laboratory services needed to obtain ASC in a timely manner. This factor could limit the applicability of an aggressive program based on obtaining ASC and preemptive placement of patients on Contact Precautions in these settings. However, with

the growing problem of antimicrobial resistance, and the recognized role of all healthcare settings for control of this problem, it is imperative that appropriate human and fiscal resources be invested to increase the feasibility of recommended control strategies in every setting.

Factors that influence selection of MDRO control measures. Although some common principles apply, the preceding literature review indicates that no single approach to the control of MDROs is appropriate for all healthcare facilities. Many factors influence the choice of interventions to be applied within an institution, including:

- ***Type and significance of problem MDROs within the institution*****.** Many facilities have an MRSA problem while others have ESBL-producing *K. pneumoniae*. Some facilities have no VRE colonization or disease; others have high rates of VRE colonization without disease; and still others have ongoing VRE outbreaks. The magnitude of the problem also varies**.** Healthcare facilities may have very low numbers of cases, e.g., with a newly introduced strain, or may have prolonged, extensive outbreaks or colonization in the population. Between these extremes, facilities may have low or high levels of endemic colonization and variable levels of infection.

- ***Population and healthcare-settings.*** The presence of high-risk patients (e.g., transplant, hematopoietic stem-cell transplant) and special-care units (e.g. adult, pediatric, and neonatal ICUs; burn; hemodialysis) will influence surveillance needs and could limit the areas of a facility targeted for MDRO control interventions. Although it appears that MDRO transmission seldom occurs in ambulatory and outpatient settings, some patient populations (e.g., hemodialysis, cystic fibrosis) and patients receiving chemotherapeutic agents are at risk for colonization and infection with MDROs. Furthermore, the emergence of VRSA within the outpatient setting(22, 23, 25) demonstrates that even these settings need to make MDRO prevention a priority.

Differences of opinion on the optimal strategy to control MDROs. Published guidance on the control of MDROs reflects areas of ongoing debate on optimal control strategies. A key issue is the use of ASC in control efforts and preemptive use of Contact Precautions pending negative surveillance culture results(107, 321, 322). The various guidelines currently available exhibit a spectrum of approaches, which their authors deem to be evidence-based. One guideline for control of MRSA and VRE, the Society for Healthcare Epidemiology of America (SHEA) guideline from 2003(107), emphasizes routine use of ASC and Contact Precautions. That position paper does not address control of MDR-GNBs. The salient features of SHEA recommendations for MRSA and VRE control and the recommendations in this guideline for control of MDROs, including MRSA and VRE, have been compared(323); recommended interventions are similar. Other guidelines for VRE and MRSA, e.g., those proffered by the Michigan Society for Infection Control (www.msic-online.org/resource_sections/aro_guidelines), emphasize consistent practice of Standard Precautions and tailoring the use of ASC and Contact Precautions to local conditions, the specific MDROs that are prevalent and being transmitted, and the presence of risk factors for transmission. A variety of approaches have reduced MDRO rates(3, 164, 165, 209, 214, 240, 269, 324). Therefore, selection of interventions for controlling MDRO transmission should be based on assessments of the local problem, the prevalence of various MDRO and feasibility. Individual facilities should seek appropriate guidance and adopt effective measures that fit their circumstances and needs. Most studies have been in acute care settings; for non-acute care settings (e.g., LCTF, small rural hospitals), the optimal approach is not well defined.

Two-Tiered Approach for Control of MDROs. Reports describing successful control of MDRO transmission in healthcare facilities have included seven categories of interventions (Table 3). As a rule, these reports indicate that facilities confronted with an MDRO problem selected a combination of control measures, implemented them, and reassessed their impact. In some cases, new measures were added serially to further enhance control efforts. This evidence indicates that the control of MDROs is a dynamic process that requires a systematic approach tailored to the problem and healthcare setting. The nature of this evidence gave rise to the two-tiered approach to MDRO control

recommended in this guideline. This approach provides the flexibility needed to prevent and control MDRO transmission in every kind of facility addressed by this guideline. Detailed recommendations for MDRO control in all healthcare settings follow and are summarized in Table 3. Table 3, which applies to all healthcare settings, contains two tiers of activities. In the first tier are the baseline level of MDRO control activities designed to ensure recognition of MDROs as a problem, involvement of healthcare administrators, and provision of safeguards for managing unidentified carriers of MDROs.

With the emergence of an MDRO problem that cannot be controlled with the basic set of infection control measures, additional control measures should be selected from the second tier of interventions presented in Table 3. Decisions to intensify MDRO control activity arise from surveillance observations and assessments of the risk to patients in various settings. Circumstances that may trigger these decisions include:

- Identification of an MDRO from even one patient in a facility or special unit with a highly vulnerable patient population (e.g., an ICU, NICU, burn unit) that had previously not encountered that MDRO.
- Failure to decrease the prevalence or incidence of a specific MDRO (e.g., incidence of resistant clinical isolates) despite infection control efforts to stop its transmission.(Statistical process control charts or other validated methods that account for normal variation can be used to track rates of targeted MDROs)(205, 325, 326).

The combination of new or increased frequency of MDRO isolates and patients at risk necessitates escalation of efforts to achieve or re-establish control, i.e., to reduce rates of transmission to the lowest possible level. Intensification of MDRO control activities should begin with an assessment of the problem and evaluation of the effectiveness of measures in current use. Once the problem is defined, appropriate additional control measures should be selected from the second tier of Table 3. A knowledgeable infection prevention and control professional or healthcare epidemiologist should make this determination. This approach requires support from the governing body and medical staff of the facility. Once interventions are implemented, ongoing surveillance should be used to determine whether selected control measures are effective and if additional measures or consultation are

indicated. The result of this process should be to decrease MDRO rates to minimum levels. Healthcare facilities must not accept ongoing MDRO outbreaks or high endemic rates as the status quo. With selection of infection control measures appropriate to their situation, all facilities *can achieve* the desired goal and reduce the MDRO burden substantially.

V. Prevention of transmission of Multidrug Resistant Organisms (Table 3)

The CDC/HICPAC system for categorizing recommendations is as follows:

Category IA Strongly recommended for implementation and strongly supported by well-designed experimental, clinical, or epidemiologic studies.

Category IB Strongly recommended for implementation and supported by some experimental, clinical, or epidemiologic studies and a strong theoretical rationale.

Category IC Required for implementation, as mandated by federal and/or state regulation or standard.

Category II Suggested for implementation and supported by suggestive clinical or epidemiologic studies or a theoretical rationale.

No recommendation Unresolved issue. Practices for which insufficient evidence or no consensus regarding efficacy exists.

V.A. General recommendations for all healthcare settings independent of the prevalence of multidrug resistant organism (MDRO) infections or the population served.

V.A.1. Administrative measures

V.A.1.a. Make MDRO prevention and control an organizational patient safety priority.(3, 146, 151, 154, 182, 185, 194, 205, 208, 210, 242, 327, 328) *Category IB*

V.A.1.b. Provide administrative support, and both fiscal and human resources, to prevent and control MDRO transmission within the healthcare organization (3, 9, 146, 152, 182-184, 208, 328, 329) *Category IB*

V.A.1.c. In healthcare facilities without expertise for analyzing epidemiologic data, recognizing MDRO problems, or devising effective control strategies (e.g., small or rural hospitals, rehabilitation centers, long-term care facilities [LTCFs], freestanding ambulatory centers), identify experts who can provide consultation as needed.(151, 188) *Category II*

V.A.1.d. Implement systems to communicate information about reportable MDROs [e.g., VRSA, VISA, MRSA, Penicillin resistant *S. pneumoniae(PRSP)*] to administrative personnel and as required by state and local health

authorities (www.cdc.gov/epo/dphsi/nndsshis.htm). Refer to websites for updated requirements of local and state health departments. *Category II/IC*

V.A.1.e. Implement a multidisciplinary process to monitor and improve healthcare personnel (HCP) adherence to recommended practices for Standard and Contact Precautions(3, 105, 182, 184, 189, 242, 273, 312, 330). *Category IB*

V.A.1.f. Implement systems to designate patients known to be colonized or infected with a targeted MDRO and to notify receiving healthcare facilities and personnel prior to transfer of such patients within or between facilities.(87, 151) *Category IB*

V.A.1.g. Support participation of the facility or healthcare system in local, regional, and national coalitions to combat emerging or growing MDRO problems.(41, 146, 151, 167, 188, 206, 207, 211, 331). *Category IB*

V.A.1.h. Provide updated feedback at least annually to healthcare providers and administrators on facility and patient-care-unit trends in MDRO infections. Include information on changes in prevalence or incidence of infection, results of assessments for system failures, and action plans to improve adherence to and effectiveness of recommended infection control practices to prevent MDRO transmission.(152, 154, 159, 184, 204, 205, 242, 312, 332) *Category IB*

V.A.2. Education and training of healthcare personnel

V.A.2.a. Provide education and training on risks and prevention of MDRO transmission during orientation and periodic educational updates for healthcare personnel; include information on organizational experience with MDROs and prevention strategies.(38, 152, 154, 173, 176, 189, 190, 203, 204, 217, 242, 330, 333, 334) *Category IB*

V.A.3. Judicious use of antimicrobial agents. The goal of the following recommendations is to ensure that systems are in place to promote optimal treatment of infections and appropriate antimicrobial use.

V.A.3.a. In hospitals and LTCFs, ensure that a multidisciplinary process is in place to review antimicrobial utilization, local susceptibility patterns

(antibiograms), and antimicrobial agents included in the formulary to foster appropriate antimicrobial use.(209, 212, 214, 215, 217, 242, 254, 334-339) *Category IB*

V.A.3.b. Implement systems (e.g., computerized physician order entry, comment in microbiology susceptibility report, notification from a clinical pharmacist or unit director) to prompt clinicians to use the appropriate antimicrobial agent and regimen for the given clinical situation.(156, 157, 161, 166, 174, 175, 212, 214, 218, 254, 334, 335, 337, 340-346) *Category IB*

V.A.3.b.i. Provide clinicians with antimicrobial susceptibility reports and analysis of current trends, updated at least annually, to guide antimicrobial prescribing practices.(342, 347) *Category IB*

V.A.3.b.ii. In settings that administer antimicrobial agents but have limited electronic communication system infrastructures to implement physician prompts (e.g., LTCFs, home care and infusion companies), implement a process for appropriate review of prescribed antimicrobials. Prepare and distribute reports to prescribers that summarize findings and provide suggestions for improving antimicrobial use. (342, 348, 349) *Category II*

V.A.4. Surveillance

V.A.4.a. In microbiology laboratories, use standardized laboratory methods and follow published guidance for determining antimicrobial susceptibility of targeted (e.g., MRSA, VRE, MDR-ESBLs) and emerging (e.g., VRSA, MDR-*Acinetobacter baumannii*) MDROs.(8, 154, 177, 190, 193, 209, 254, 347, 350-353) *Category IB*

V.A.4.b. In all healthcare organizations, establish systems to ensure that clinical microbiology laboratories (in-house and out-sourced) promptly notify infection control staff or a medical director/ designee when a novel resistance pattern for that facility is detected.(9, 22, 154, 162, 169) *Category IB*

V.A.4.c. In hospitals and LTCFs, develop and implement laboratory protocols for storing isolates of selected MDROs for molecular typing when needed to

confirm transmission or delineate the epidemiology of the MDRO within the healthcare setting.(7, 8, 38, 140, 153, 154, 187, 190, 208, 217, 354, 355) *Category IB*

V.A.4.d. Prepare facility-specific antimicrobial susceptibility reports as recommended by the Clinical and Laboratory Standards Institute (CLSI) (www.phppo.cdc.gov/dls/master/default.aspx); monitor these reports for evidence of changing resistance patterns that may indicate the emergence or transmission of MDROs.(347, 351, 356, 357) *Category IB/IC*

V.A.4.d.i. In hospitals and LTCFs with special-care units (e.g., ventilator-dependent, ICU, or oncology units), develop and monitor unit-specific antimicrobial susceptibility reports.(358-361) *Category IB*

V.A.4.d.ii. Establish a frequency for preparing summary reports based on volume of clinical isolates, with updates at least annually.(347, 362) *Category II/IC*

V.A.4.d.iii. In healthcare organizations that outsource microbiology laboratory services (e.g., ambulatory care, home care, LTCFs, smaller acute care hospitals), specify by contract that the laboratory provide either facility-specific susceptibility data or local or regional aggregate susceptibility data in order to identify prevalent MDROs and trends in the geographic area served.(363) *Category II*

V.A.4.e. Monitor trends in the incidence of target MDROs in the facility over time using appropriate statistical methods to determine whether MDRO rates are decreasing and whether additional interventions are needed.(152, 154, 183, 193, 205, 209, 217, 242, 300, 325, 326, 364, 365) *Category IA*

V.A.4.e.i. Specify isolate origin (i.e., location and clinical service) in MDRO monitoring protocols in hospitals and other large multi-unit facilities with high-risk patients.(8, 38, 152-154, 217, 358, 361) *Category IB*

V.A.4.e.ii. Establish a baseline (e.g., incidence) for targeted MDRO isolates by reviewing results of clinical cultures; if more timely or localized information is needed, perform baseline point prevalence studies of colonization in high-risk units. When possible, distinguish

colonization from infection in analysis of these data.(152, 153, 183, 184, 189, 190, 193, 205, 242, 365) *Category IB*

V.A.5. Infection control precautions to prevent transmission of MDROs

V.A.5.a. Follow Standard Precautions during all patient encounters in all settings in which healthcare is delivered.(119, 164, 255, 315, 316) *Category IB*

V.A.5.b. Use masks according to Standard Precautions when performing splash-generating procedures (e.g., wound irrigation, oral suctioning, intubation); when caring for patients with open tracheostomies and the potential for projectile secretions; and in circumstances where there is evidence of transmission from heavily colonized sources (e.g., burn wounds). Masks are not otherwise recommended for prevention of MDRO transmission from patients to healthcare personnel during routine care (e.g., upon room entry).(8, 22, 151, 152, 154, 189, 190, 193, 208, 240, 366) *Category IB*

V.A.5.c. Use of Contact Precautions

V.A.5.c.i. In *acute-care hospitals*, implement Contact Precautions routinely for all patients infected with target MDROs and for patients that have been previously identified as being colonized with target MDROs (e.g., patients transferred from other units or facilities who are known to be colonized). (11, 38, 68, 114, 151, 183, 188, 204, 217, 242, 304) *Category IB*

V.A.5.c.ii. In LTCFs, consider the individual patient's clinical situation and prevalence or incidence of MDRO in the facility when deciding whether to implement or modify Contact Precautions in addition to Standard Precautions for a patient infected or colonized with a target MDRO. *Category II*

V.A.5.c.ii.1. For relatively healthy residents (e.g., mainly independent) follow Standard Precautions, making sure that gloves and gowns are used for contact with uncontrolled secretions, pressure ulcers, draining wounds, stool incontinence, and ostomy tubes/bags. (78-80, 85, 151, 367, 368) *Category II*

V.A.5.c.ii.2. For ill residents (e.g., those totally dependent upon healthcare personnel for healthcare and activities of daily living, ventilator-dependent) and for those residents whose infected secretions or drainage cannot be contained, use Contact Precautions in addition to Standard Precautions.(316, 369, 370) *Category II*

V.A.5.c.iii. For MDRO colonized or infected patients without draining wounds, diarrhea, or uncontrolled secretions, establish ranges of permitted ambulation, socialization, and use of common areas based on their risk to other patients and on the ability of the colonized or infected patients to observe proper hand hygiene and other recommended precautions to contain secretions and excretions.(151, 163, 371) *Category II*

V.A.5.d. In *ambulatory settings*, use Standard Precautions for patients known to be infected or colonized with target MDROs, making sure that gloves and gowns are used for contact with uncontrolled secretions, pressure ulcers, draining wounds, stool incontinence, and ostomy tubes and bags. *Category II*

V.A.5.e. In *home care settings*

- Follow Standard Precautions making sure to use gowns and gloves for contact with uncontrolled secretions, pressure ulcers, draining wounds, stool incontinence, and ostomy tubes and bags. *Category II*
- Limit the amount of reusable patient-care equipment that is brought into the home of patients infected or colonized with MDROs. When possible, leave patient-care equipment in the home until the patient is discharged from home care services. *Category II*
- If noncritical patient-care equipment (e.g., stethoscopes) cannot remain in the home, clean and disinfect items before removing them from the home, using a low to intermediate level disinfectant, or place reusable items in a plastic bag for transport

to another site for subsequent cleaning and disinfection. *Category II*

V.A.5.e.i. No recommendation is made for routine use of gloves, gowns, or both to prevent MDRO transmission in ambulatory or home care settings. *Unresolved issue*

V.A.5.e.ii. In *hemodialysis units,* follow the "Recommendations to Prevent Transmission of Infections in Chronic Hemodialysis Patients"(372)(www.cms.hhs.gov/home/regsguidance.asp). *Category IC*

V.A.5.f. Discontinuation of Contact Precautions. No recommendation can be made regarding when to discontinue Contact Precautions. *Unresolved issue* (See Background for discussion of options)

V.A.5.g. Patient placement in hospitals and LTCFs

V.A.5.g.i. When single-patient rooms are available, assign priority for these rooms to patients with known or suspected MDRO colonization or infection. Give highest priority to those patients who have conditions that may facilitate transmission, e.g., uncontained secretions or excretions.(8, 38, 110, 151, 188, 208, 240, 304) *Category IB*

V.A.5.g.ii. When single-patient rooms are not available, cohort patients with the same MDRO in the same room or patient-care area.(8, 38, 92, 151-153, 162, 183, 184, 188, 217, 242, 304) *Category IB*

V.A.5.g.iii. When cohorting patients with the same MDRO is not possible, place MDRO patients in rooms with patients who are at low risk for acquisition of MDROs and associated adverse outcomes from infection and are likely to have short lengths of stay. *Category II*

V.A.6. Environmental measures

V.A.6.a. Clean and disinfect surfaces and equipment that may be contaminated with pathogens, including those that are in close proximity to the patient (e.g., bed rails, over bed tables) and frequently-touched surfaces in the patient care environment (e.g., door knobs, surfaces in and surrounding toilets in patients' rooms) on a more frequent schedule compared to that for minimal

touch surfaces (e.g., horizontal surfaces in waiting rooms).(111, 297, 373) *Category IB*

V.A.6.b. Dedicate noncritical medical items to use on individual patients known to be infected or colonized with MDROs.(38, 217, 324, 374, 375) *Category IB*

V.A.6.c. Prioritize room cleaning of patients on Contact Precautions. Focus on cleaning and disinfecting frequently touched surfaces (e.g., bedrails, bedside commodes, bathroom fixtures in the patient's room, doorknobs) and equipment in the immediate vicinity of the patient.(109, 110, 114-117, 297, 301, 373, 376, 377) *Category IB*

V.B. Intensified interventions to prevent MDRO transmission

The interventions presented below have been utilized in various combinations to reduce transmission of MDROs in healthcare facilities. Neither the effectiveness of individual components nor that of specific combinations of control measures has been assessed in controlled trials. Nevertheless, various combinations of control elements selected under the guidance of knowledgeable content experts have repeatedly reduced MDRO transmission rates in a variety of healthcare settings.

V.B.1. Indications and approach

V.B.1.a. Indications for intensified MDRO control efforts (VII.B.1.a.i and VII.B.1.a.ii) should result in selection and implementation of one or more of the interventions described in VII.B.2 to VII.B.8 below. Individualize the selection of control measures according to local considerations(8, 11, 38, 68, 114, 152-154, 183-185, 189, 190, 193, 194, 209, 217, 242, 312, 364, 365). *Category IB*

V.B.1.a.i. When incidence or prevalence of MDROs are not decreasing despite implementation of and correct adherence to the routine control measures described above, intensify MDRO control efforts by adopting one or more of the interventions described below.(92, 152, 183, 184, 193, 365) *Category IB*

V.B.1.a.ii. When the *first* case or outbreak of an epidemiologically important MDRO (e.g., VRE, MRSA, VISA, VRSA, MDR-GNB) is identified

within a healthcare facility or unit.(22, 23, 25, 68, 170, 172, 184, 240, 242, 378) *Category IB*

V.B.1.b. Continue to monitor the incidence of target MDRO infection and colonization after additional interventions are implemented. If rates do not decrease, implement more interventions as needed to reduce MDRO transmission.(11, 38, 68, 92, 152, 175, 184, 365) *Category IB*

V.B.2. Administrative measures

V.B.2.a. Identify persons with experience in infection control and the epidemiology of MDRO, either in house or through outside consultation, for assessment of the local MDRO problem and for the design, implementation, and evaluation of appropriate control measures (3, 68, 146, 151-154, 167, 184, 190, 193, 242, 328, 377). *Category IB*

V.B.2.b. Provide necessary leadership, funding, and day-to-day oversight to implement interventions selected. Involve the governing body and leadership of the healthcare facility or system that have organizational responsibility for this and other infection control efforts.(8, 38, 152, 154, 184, 189, 190, 208) *Category IB*

V.B.2.c. Evaluate healthcare system factors for their role in creating or perpetuating transmission of MDROs, including: staffing levels, education and training, availability of consumable and durable resources, communication processes, policies and procedures, and adherence to recommended infection control measures (e.g., hand hygiene and Standard or Contact Precautions). Develop, implement, and monitor action plans to correct system failures.(3, 8, 38, 152, 154, 172, 173, 175, 188, 196, 198, 199, 208, 217, 280, 324, 379, 380) *Category IB*

V.B.2.d. During the process, update healthcare providers and administrators on the progress and effectiveness of the intensified interventions. Include information on changes in prevalence, rates of infection and colonization; results of assessments and corrective actions for system failures; degrees of adherence to recommended practices; and action plans to improve

adherence to recommended infection control practices to prevent MDRO transmission.(152, 154, 159, 184, 204, 205, 312, 332, 381) *Category IB*

V.B.3. Educational interventions

Intensify the frequency of MDRO educational programs for healthcare personnel, especially those who work in areas in which MDRO rates are not decreasing. Provide individual or unit-specific feedback when available.(3, 38, 152, 154, 159, 170, 182, 183, 189, 190, 193, 194, 204, 205, 209, 215, 218, 312) *Category IB*

V.B.4. Judicious use of antimicrobial agents

Review the role of antimicrobial use in perpetuating the MDRO problem targeted for intensified intervention. Control and improve antimicrobial use as indicated. Antimicrobial agents that may be targeted include vancomycin, third-generation cephalosporins, and anti-anaerobic agents for VRE(217); third-generation cephalosporins for ESBLs(212, 214, 215); and quinolones and carbapenems(80, 156, 166, 174, 175, 209, 218, 242, 254, 329, 334, 335, 337, 341). *Category IB*

V.B.5. Surveillance

V.B.5.a. Calculate and analyze prevalence and incidence rates of targeted MDRO infection and colonization in populations at risk; when possible, distinguish colonization from infection(152, 153, 183, 184, 189, 190, 193, 205, 215, 242, 365). *Category IB*

V.B.5.a.i. Include only one isolate per patient, not multiple isolates from the same patient, when calculating rates(347, 382). *Category II*

V.B.5.a.ii. Increase the frequency of compiling and monitoring antimicrobial susceptibility summary reports for a targeted MDRO as indicated by an increase in incidence of infection or colonization with that MDRO. C*ategory II*

V.B.5.b. Develop and implement protocols to obtain active surveillance cultures (ASC) for targeted MDROs from patients in populations at risk (e.g., patients in intensive care, burn, bone marrow/stem cell transplant, and oncology units; patients transferred from facilities known to have high

MDRO prevalence rates; roommates of colonized or infected persons; and patients known to have been previously infected or colonized with an MDRO).(8, 38, 68, 114, 151-154, 167, 168, 183, 184, 187-190, 192, 193, 217, 242) *Category IB*

V.B.5.b.i. Obtain ASC from areas of skin breakdown and draining wounds. In addition, include the following sites according to target MDROs:

V.B.5.b.i.1. For MRSA: Sampling the anterior nares is usually sufficient; throat, endotracheal tube aspirate, percutaneous gastrostomy sites, and perirectal or perineal cultures may be added to increase the yield. Swabs from several sites may be placed in the same selective broth tube prior to transport.(117, 383, 384) *Category IB*

V.B.5.b.i.2. For VRE: Stool, rectal, or perirectal samples should be collected.(154, 193, 217, 242)
Category IB

V.B.5.b.i.3. For MDR-GNB: Endotracheal tube aspirates or sputum should be cultured if a respiratory tract reservoir is suspected, (e.g., *Acinetobacter* spp., *Burkholderia* spp.).(385, 386) *Category IB*.

V.B.5.b.ii. Obtain surveillance cultures for the target MDRO from patients at the time of admission to high-risk areas, e.g., ICUs, and at periodic intervals as needed to assess MDRO transmission.(8, 151, 154, 159, 184, 208, 215, 242, 387) *Category IB*

V.B.5.c. Conduct culture surveys to assess the efficacy of the enhanced MDRO control interventions.

V.B.5.c.i. Conduct serial (e.g., weekly, until transmission has ceased and then decreasing frequency) unit-specific point prevalence culture surveys of the target MDRO to determine if transmission has decreased or ceased.(107, 167, 175, 184, 188, 218, 339) *Category IB*

V.B.5.c.ii. Repeat point-prevalence culture surveys at routine intervals or at time of patient discharge or transfer until transmission has ceased.(8, 152-154, 168, 178, 190, 215, 218, 242, 388) *Category IB*

V.B.5.c.iii. If indicated by assessment of the MDRO problem, collect cultures to asses the colonization status of roommates and other patients with substantial exposure to patients with known MDRO infection or colonization.(25, 68, 167, 193) *Category IB*

V.B.5.d. Obtain cultures of healthcare personnel for target MDRO when there is epidemiologic evidence implicating the healthcare staff member as a source of ongoing transmission.(153, 365) *Category IB*

V.B.6. Enhanced infection control precautions

V.B.6.a. Use of Contact Precautions

V.B.6.a.i. Implement Contact Precautions routinely for all patients colonized or infected with a target MDRO.(8, 11, 38, 68, 114, 151, 154, 183, 188, 189, 217, 242, 304) *Category IA*

V.B.6.a.ii. Because environmental surfaces and medical equipment, especially those in close proximity to the patient, may be contaminated, don gowns and gloves *before or upon entry* to the patient's room or cubicle.(38, 68, 154, 187, 189, 242) *Category IB*

V.B.6.a.iii. In LTCFs, modify Contact Precautions to allow MDRO-colonized/infected patients whose site of colonization or infection can be appropriately contained and who can observe good hand hygiene practices to enter common areas and participate in group activities.(78, 86, 151, 367) *Category IB*

V.B.6.b. When ASC are obtained as part of an intensified MDRO control program, implement Contact Precautions until the surveillance culture is reported negative for the target MDRO.(8, 30, 153, 389, 390) C*ategory IB*

V.B.6.c. No recommendation is made regarding universal use of gloves, gowns, or both in high-risk units in acute-care hospitals.(153, 273, 312, 320, 391) *Unresolved issue*

V.B.7. Implement policies for patient admission and placement as needed to prevent transmission of a problem MDRO.(183, 184, 189, 193, 242, 339, 392) *Category IB*

V.B.7.a.i. Place MDRO patients in single-patient rooms.(6, 151, 158, 160, 166, 170, 187, 208, 240, 282, 393-395) *Category IB*

V.B.7.a.ii. Cohort patients with the same MDRO in designated areas (e.g., rooms, bays, patient care areas.(8, 151, 152, 159, 161, 176, 181, 183, 184, 188, 208, 217, 242, 280, 339, 344) *Category IB*

V.B.7.a.iii. When transmission continues despite adherence to Standard and Contact Precautions and cohorting patients, assign dedicated nursing and ancillary service staff to the care of MDRO patients only. Some facilities may consider this option when intensified measures are first implemented.(184, 217, 242, 278) *Category IB*

V.B.7.a.iv. Stop new admissions to the unit of facility if transmission continues despite the implementation of the enhanced control measures described above. (Refer to state or local regulations that may apply upon closure of hospital units or services.).(9, 38, 146, 159, 161, 168, 175, 205, 279, 280, 332, 339, 396) *Category IB*

V.B.8. Enhanced environmental measures

V.B.8.a. Implement patient-dedicated or single-use disposable noncritical equipment (e.g., blood pressure cuff, stethoscope) and instruments and devices.(38, 104, 151, 156, 159, 163, 181, 217, 324, 329, 367, 389, 390, 394) *Category IB*

V.B.8.b. Intensify and reinforce training of environmental staff who work in areas targeted for intensified MDRO control and monitor adherence to environmental cleaning policies. Some facilities may choose to assign dedicated staff to targeted patient care areas to enhance consistency of proper environmental cleaning and disinfection services.(38, 154, 159, 165, 172, 173, 175, 178-181, 193, 205, 208, 217, 279, 301, 327, 339, 397) *Category IB*

V.B.8.c. Monitor (i.e., supervise and inspect) cleaning performance to ensure consistent cleaning and disinfection of surfaces in close proximity to the patient and those likely to be touched by the patient and HCP (e.g.,

bedrails, carts, bedside commodes, doorknobs, faucet handles).(8, 38, 109, 111, 154, 169, 180, 208, 217, 301, 333, 398) *Category IB*

V.B.8.d. Obtain environmental cultures (e.g., surfaces, shared medical equipment) when there is epidemiologic evidence that an environmental source is associated with ongoing transmission of the targeted MDRO.(399-402) *Category IB*

V.B.8.e. Vacate units for environmental assessment and intensive cleaning when previous efforts to eliminate environmental reservoirs have failed.(175, 205, 279, 339, 403) *Category II*

V.B.9. Decolonization

V.B.9.a. Consult with physicians with expertise in infectious diseases and/or healthcare epidemiology on a case-by-case basis regarding the appropriate use of decolonization therapy for patients or staff during limited periods of time, as a component of an intensified MRSA control program).(152, 168, 170, 172, 183, 194, 304) *Category II*

V.B.9.b. When decolonization for MRSA is used, perform susceptibility testing for the decolonizing agent against the target organism in the individual being treated or the MDRO strain that is epidemiologically implicated in transmission. Monitor susceptibility to detect emergence of resistance to the decolonizing agent. Consult with a microbiologist for appropriate testing for mupirocin resistance, since standards have not been established.(289, 290, 304, 308) *Category IB*

V.B.9.b.i. Because mupirocin-resistant strains may emerge and because it is unusual to eradicate MRSA when multiple body sites are colonized, do not use topical mupirocin *routinely* for MRSA decolonization of patients as a component of MRSA control programs in any healthcare setting.(289, 404) *Category IB*

V.B.9.b.ii. Limit decolonization of HCP found to be colonized with MRSA to persons who have been epidemiologically linked as a likely source of ongoing transmission to patients. Consider reassignment of HCP

if decolonization is not successful and ongoing transmission to patients persists.(120, 122, 168) *Category IB*

V.B.9.c. No recommendation can be made for decolonizing patients with VRE or MDR-GNB. Regimens and efficacy of decolonization protocols for VRE and MDR-GNB have not been established.(284, 286, 288, 307, 387, 405) *Unresolved issue*

Glossary - Multidrug-Resistant Organisms

Ambulatory care settings. Facilities that provide health care to patients who do not remain overnight (e.g., hospital-based outpatient clinics, nonhospital-based clinics and physician offices, urgent care centers, surgicenters, free-standing dialysis centers, public health clinics, imaging centers, ambulatory behavioral health and substance abuse clinics, physical therapy and rehabilitation centers, and dental practices.

Cohorting. In the context of this guideline, this term applies to the practice of grouping patients infected or colonized with the same infectious agent together to confine their care to one area and prevent contact with susceptible patients (cohorting patients). During outbreaks, healthcare personnel may be assigned to a cohort of patients to further limit opportunities for transmission (cohorting staff).

Contact Precautions. Contact Precautions are a set of practices used to prevent transmission of infectious agents that are spread by direct or indirect contact with the patient or the patient's environment. Contact Precautions also apply where the presence of excessive wound drainage, fecal incontinence, or other discharges from the body suggest an increased transmission risk. A single patient room is preferred for patients who require Contact Precautions. When a single patient room is not available, consultation with infection control is helpful to assess the various risks associated with other patient placement options (e.g., cohorting, keeping the patient with an existing roommate). In multi-patient rooms, $\geq$3 feet spatial separation of between beds is advised to reduce the opportunities for inadvertent sharing of items between the infected/colonized patient and other patients. Healthcare personnel caring for patients on Contact Precautions wear a gown and gloves for all interactions that may involve contact with the patient or potentially contaminated areas in the patient's environment. Donning of gown and gloves upon room entry, removal before exiting the patient room and performance of hand hygiene immediately upon exiting are done to contain pathogens.

Epidemiologically important pathogens. Infectious agents that have one or more of the following characteristics: 1)A propensity for transmission within healthcare facilities based on published reports and the occurrence of temporal or geographic clusters of $\geq$ 2 patients, (e.g., VRE, MRSA and MSSA, *Clostridium difficile*, norovirus, RSV, influenza, rotavirus, *Enterobacter* spp; *Serratia* spp., group A streptococcus). However, for group A streptococcus, most experts consider a single case of healthcare-associated disease a trigger for investigation and enhanced control measures because of the devastating outcomes associated with HAI group A streptococcus infections. For susceptible bacteria that are known to be associated with asymptomatic colonization, isolation from normally sterile body fluids in patients with significant clinical disease would be the trigger to consider the organism as epidemiologically important. 2) Antimicrobial resistance implications:

- Resistance to first-line therapies (e.g., MRSA, VRE, VISA, VRSA, ESBL-producing organisms).
- Unusual or usual agents with unusual patterns of resistance within a facility, (e.g., the first isolate of *Burkholderia cepacia* complex or *Ralstonia* spp. in non-CF patients or a quinolone-resistant strain of Pseudomonas in a facility.
- Difficult to treat because of innate or acquired resistance to multiple classes of antimicrobial agents (e.g., *Stenotrophomonas maltophilia, Acinetobacter spp.*).

3) Associated with serious clinical disease, increased morbidity and mortality (e.g., MRSA and MSSA, group A streptococcus); or 4) A newly discovered or reemerging pathogen. The strategies described for MDROs may be applied for control of epidemiologically important organisms other than MDROs.

Hand hygiene. A general term that applies to any one of the following: 1) handwashing with plain (nonantimicrobial) soap and water); 2) antiseptic hand wash (soap containing antiseptic agents and water); 3) antiseptic hand rub (waterless antiseptic product, most often alcohol-based, rubbed on all surfaces of hands); or 4) surgical hand antisepsis

(antiseptic hand wash or antiseptic hand rub performed preoperatively by surgical personnel to eliminate transient hand flora and reduce resident hand flora).

Healthcare-associated infection (HAI). An infection that develops in a patient who is cared for in any setting where healthcare is delivered (e.g., acute care hospital, chronic care facility, ambulatory clinic, dialysis center, surgicenter, home) and is related to receiving health care (i.e., was not incubating or present at the time healthcare was provided). In ambulatory and home settings, HAI would apply to any infection that is associated with a medical or surgical intervention performed in those settings.

Healthcare epidemiologist A person whose primary training is medical (M.D., D.O.) and/or masters or doctorate-level epidemiology who has received advanced training in healthcare epidemiology. Typically these professionals direct or provide consultation to an infection prevention and control program in a hospital, long term care facility (LTCF), or healthcare delivery system (also see infection prevention and control professional).

Healthcare personnel (HCP). All paid and unpaid persons who work in a healthcare setting, also known as healthcare workers (e.g. any person who has professional or technical training in a healthcare-related field and provides patient care in a healthcare setting or any person who provides services that support the delivery of healthcare such as dietary, housekeeping, engineering, maintenance personnel).

Home care. A wide-range of medical, nursing, rehabilitation, hospice, and social services delivered to patients in their place of residence (e.g., private residence, senior living center, assisted living facility). Home health-care services include care provided by home health aides and skilled nurses, respiratory therapists, dieticians, physicians, chaplains, and volunteers; provision of durable medical equipment; home infusion therapy; and physical, speech, and occupational therapy.

Infection prevention and control professional (ICP). A person whose primary training is in either nursing, medical technology, microbiology, or epidemiology and who has acquired

specialized training in infection control. Responsibilities may include collection, analysis, and feedback of infection data and trends to healthcare providers; consultation on infection risk assessment, prevention and control strategies; performance of education and training activities; implementation of evidence-based infection control practices or those mandated by regulatory and licensing agencies; application of epidemiologic principles to improve patient outcomes; participation in planning renovation and construction projects (e.g., to ensure appropriate containment of construction dust); evaluation of new products or procedures on patient outcomes; oversight of employee health services related to infection prevention; implementation of preparedness plans; communication within the healthcare setting, with local and state health departments, and with the community at large concerning infection control issues; and participation in research.

***Infection prevention and control program*.** A multidisciplinary program that includes a group of activities to ensure that recommended practices for the prevention of healthcare-associated infections are implemented and followed by healthcare personnel, making the healthcare setting safe from infection for patients and healthcare personnel. The Joint Commission on Accreditation of Healthcare Organizations (JCAHO) requires the following five components of an infection prevention and control program for accreditation: 1) *surveillance*: monitoring patients and healthcare personnel for acquisition of infection and/or colonization; 2) *investigation:* identification and analysis of infection problems or undesirable trends; 3) *prevention*: implementation of measures to prevent transmission of infectious agents and to reduce risks for device- and procedure-related infections; 4) *control*: evaluation and management of outbreaks; and 5) *reporting:* provision of information to external agencies as required by state and federal law and regulation (www.jcaho.org). The infection prevention and control program staff has the ultimate authority to determine infection control policies for a healthcare organization with the approval of the organization's governing body.

Long-term care facilities (LTCFs).An array of residential and outpatient facilities designed to meet the bio-psychosocial needs of persons with sustained self-care deficits. These include skilled nursing facilities, chronic disease hospitals, nursing homes, foster and group homes, institutions for the developmentally disabled, residential care facilities, assisted

living facilities, retirement homes, adult day health care facilities, rehabilitation centers, and long-term psychiatric hospitals.

Mask. A term that applies collectively to items used to cover the nose and mouth and includes both procedure masks and surgical masks (www.fda.gov/cdrh/ode/guidance/094.html#4).

Multidrug-resistant organisms (MDROs). In general, bacteria (excluding *M. tuberculosis)* that are resistant to one or more classes of antimicrobial agents and usually are resistant to all but one or two commercially available antimicrobial agents (e.g., MRSA, VRE, extended spectrum beta-lactamase [ESBL]-producing or intrinsically resistant gram-negative bacilli).

Nosocomial infection. Derived from two Greek words “nosos” (disease) and “komeion” (to take care of). Refers to any infection that develops during or as a result of an admission to an acute care facility (hospital) and was not incubating at the time of admission.

Standard Precautions*.* A group of infection prevention practices that apply to all patients, regardless of suspected or confirmed diagnosis or presumed infection status. Standard Precautions are a combination and expansion of Universal Precautions and Body Substance Isolation. Standard Precautions are based on the principle that all blood, body fluids, secretions, excretions except sweat, nonintact skin, and mucous membranes may contain transmissible infectious agents. Standard Precautions includes hand hygiene, and depending on the anticipated exposure, use of gloves, gown, mask, eye protection, or face shield. Also, equipment or items in the patient environment likely to have been contaminated with infectious fluids must be handled in a manner to prevent transmission of infectious agents, (e.g. wear gloves for handling, contain heavily soiled equipment, properly clean and disinfect or sterilize reusable equipment before use on another patient).

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Table 1. Categorization of Reports about Control of MDROs in Healthcare Settings, 1982-2005

MDRO	MDR-GNB	MRSA	VRE
No. of Studies Reviewed/category	30	35	39
Types of Healthcare Facilities from which Study or Report Arose			
No. (%) from academic facilities[α]	30 (100)	28 (80)	33 (85)
No. (%) from other hospitals	0	4 (11)	3 (8)
No. (%) from LTCFs	0	1 (3)	2 (5)
No. (%) from multiple facilities in a region	0	2 (6)	1 (2)
Unit of Study for MDRO Control Efforts			
Special unit[β]	20	13	19
Hospital	10	19	17
LTCF	0	1	2
Region	0	2	1
Nature of Study or Report on MDRO Control[χ]			
Outbreak	22	19	28
Non-outbreak	8	16	11
Total Period of Observation after Interventions Introduced			
Less than 1 year	17	14	25
1-2 years	6	6	6
2-5 years	5	11	8
Greater than 5 years	2	4	
Numbers of Control Measures Employed in Outbreaks/Studies			
Range	2-12	0-11	1-12
Median	7	7	8
Mode	8	7	9

[α] Variably described as university hospitals, medical school affiliated hospitals, VA teaching hospitals, and, to a much lesser extent, community teaching hospitals

[β] Includes intensive care units, burn units, dialysis units, hematology/oncology units, neonatal units, neonatal intensive care units, and, in a few instances, individual wards of a hospital

[χ] Based on authors' description – if they called their experience an outbreak or not; authors vary in use of term so there is probable overlap between two categories

Table 2. Control Measures for MDROs Employed in Studies Performed in Healthcare Settings, 1982-2005

Focus of MDRO (No. of Studies)	MDR-GNB (n=30)	MRSA (n=35)	VRE (n=39)
	No. (%) of Studies Using Control Measure		
Education of staff, patients or visitors	19 (63)	11 (31)	20 (53)
Emphasis on handwashing	16 (53)	21 (60)	9 (23)
Use of antiseptics for handwashing	8 (30)	12 (36)	16 (41)
Contact Precautions or glove use[α]	20 (67)	27 (77)	34 (87)
Private Rooms	4 (15)	10 (28)	10 (27)
Segregation of cases	4 (15)	3 (9)	5 (14)
Cohorting of Patients	11 (37)	12 (34)	14 (36)
Cohorting of Staff	2 (7)	6 (17)	9 (23)
Change in Antimicrobial Use	12 (41)	1 (3)	17 (44)
Surveillance cultures of patients	19 (63)	34 (97)	36 (92)
Surveillance cultures of staff	9 (31)	8 (23)	7 (19)
Environmental cultures	15 (50)	14 (42)	15 (38)
Extra cleaning & disinfection	11 (37)	7 (21)	20 (51)
Dedicated Equipment	5 (17)	0	12 (32)
Decolonization	3 (10)	25 (71)	4 (11)
Ward closure to new admission or to all patients	6 (21)	4 (12)	5 (14)
Other miscellaneous measures	6 (22) [β]	9 (27)[χ]	17 (44)[δ]

[α] Contact Precautions mentioned specifically, use of gloves with gowns or aprons mentioned, barrier precautions, strict isolation, all included under this heading

[β] includes signage, record flagging, unannounced inspections, selective decontamination, and peer compliance monitoring (1 to 4 studies employing any of these measures)

[χ] includes requirements for masks, signage, record tracking, alerts, early discharge, and preventive isolation of new admissions pending results of screening cultures (1 to 4 studies employing any of these measures)

[δ] includes computer flags, signage, requirement for mask, one-to-one nursing, changing type of thermometer used, and change in rounding sequence (1 to 7 studies employing any of these measures)

References for Tables 1 and 2

MDR-GNBs: (6, 8, 9, 11, 16, 38, 174, 175, 180, 209, 210, 213-215, 218, 334, 388, 406, 407)

MRSA: (68, 89, 152, 153, 165-173, 183, 188, 194, 204, 205, 208, 240, 269, 279, 280, 289, 304, 312, 327, 365, 392, 397, 408-412)

Table 3.

Tier 1. General Recommendations for Routine Prevention and Control of MDROs in Healthcare Settings

Administrative Measures/Adherence Monitoring	MDRO Education	Judicious Antimicrobial Use	Surveillance	Infection Control Precautions to Prevent Transmission	Environmental Measures	Decolonization
Make MDRO prevention/control an organizational priority. Provide administrative support and both fiscal and human resources to prevent and control MDRO transmission. *(IB)* Identify experts who can provide consultation and expertise for analyzing epidemiologic data, recognizing MDRO problems, or devising effective control strategies, as needed. *(II)* **Implement systems to communicate information about reportable MDROs to administrative personnel and state/local health departments. *(II)*** Implement a multi-disciplinary process to monitor and improve HCP adherence to recommended practices for Standard and Contact Precautions.*(IB)* Implement systems to designate patients known to be colonized or infected with a targeted MDRO and to notify receiving healthcare facilities or personnel prior to transfer of such patients within or between facilities. *(IB)* Support participation in local, regional and/or national coalitions to combat emerging or growing MDRO problems.*(IB)* Provide updated feedback at least annually to healthcare providers and administrators on facility and patient-care unit MDRO infections. Include information on changes in prevalence and incidence, problem assessment and performance improvement plans. (*IB)*	Provide education and training on risks and prevention of MDRO transmission during orientation and periodic educational updates for HCP; include information on organizational experience with MDROs and prevention strategies. *(IB)*	In hospitals and LTCFs, ensure that a multi-disciplinary process is in place to review local susceptibility patterns (antibiograms), and antimicrobial agents included in the formulary, to foster appropriate antimicrobial use. *(IB)* Implement systems (e.g., CPOE, susceptibility report comment, pharmacy or unit director notification) to prompt clinicians to use the appropriate agent and regimen for the given clinical situation. *(IB)* Provide clinicians with antimicrobial susceptibility reports and analysis of current trends, updated at least annually, to guide antimicrobial prescribing practices. *(IB)* In settings with limited electronic communication system infrastructures to implement physician prompts, etc., at a minimum implement a process to review antibiotic use. Prepare and distribute reports to providers. *(II)*	Use standardized laboratory methods and follow published guidelines for determining antimicrobial susceptibilities of targeted and emerging MDROs. Establish systems to ensure that clinical micro labs (in-house and outsourced) promptly notify infection control or a medical director/designee when a novel resistance pattern for that facility is detected. *(IB)* In hospitals and LTCFs: …develop and implement laboratory protocols for storing isolates of selected MDROs for molecular typing when needed to confirm transmission or delineate epidemiology of MDRO in facility. *(IB)* …establish laboratory-based systems to detect and communicate evidence of MDROs in clinical isolates *(IB)* …prepare facility-specific antimicrobial susceptibility reports as recommended by CLSI; monitor reports for evidence of changing resistance that may indicate emergence or transmission of MDROs *(IA/IC)* …develop and monitor special-care unit-specific antimicrobial susceptibility reports (e.g., ventilator-dependent units, ICUs, oncology units). (*IB)* …monitor trends in incidence of target MDROs in the facility over time to determine if MDRO rates are decreasing or if additional interventions are needed. (*IA)*	Follow Standard Precautions in all healthcare settings. *(IB)* Use of Contact Precautions (CP): --- In *acute care settings* : Implement CP for all patients known to be colonized/infected with target MDROs.*(IB)* --- In *LTCFs: Consider the individual patient's clinical situation and facility resources in deciding whether to implement CP (II)* --- In *ambulatory and home care settings, follow Standard Precautions (II)* ---In *hemodialysis units*: Follow dialysis specific guidelines *(IC)* No recommendation can be made regarding when to discontinue CP. (*Unresolved issue)* Masks are not recommended for routine use to prevent transmission of MDROs from patients to HCWs. Use masks according to Standard Precautions when performing splash-generating procedures, caring for patients with open tracheostomies with potential for projectile secretions, and when there is evidence for transmission from heavily colonized sources (e.g., burn wounds). Patient placement in hospitals and LTCFs: When single-patient rooms are available, assign priority for these rooms to patients with known or suspected MDRO colonization or infection. Give highest priority to those patients who have conditions that may facilitate transmission, e.g., uncontained secretions or excretions. When single-patient rooms are not available, cohort patients with the same MDRO in the same room or patient-care area. *(IB)* When cohorting patients with the same MDRO is not possible, place MDRO patients in rooms with patients who are at low risk for acquisition of MDROs and associated adverse outcomes from infection and are likely to have short lengths of stay. (*II)*	Follow recommended cleaning, disinfection and sterilization guidelines for maintaining patient care areas and equipment. Dedicate non-critical medical items to use on individual patients known to be infected or colonized with an MDRO. Prioritize room cleaning of patients on Contact Precautions. Focus on cleaning and disinfecting frequently touched surfaces (e.g., bed rails, bedside commodes, bathroom fixtures in patient room, doorknobs) and equipment in immediate vicinity of patient.	Not recommended routinely

Tier 2. Recommendations for Intensified MDRO control efforts

Institute one or more of the interventions described below when 1) incidence or prevalence of MDROs are not decreasing despite the use of routine control measures; or 2) the *first* case or outbreak of an epidemiologically important MDRO (e.g., VRE, MRSA, VISA, VRSA, MDR-GNB) is identified within a healthcare facility or unit (*IB)* Continue to monitor the incidence of target MDRO infection and colonization; if rates do not decrease, implement additional interventions as needed to reduce MDRO transmission.

Administrative Measures/Adherence Monitoring	**MDRO Education**	**Judicious Antimicrobial Use**	**Surveillance**	Infection Control Precautions to Prevent Transmission	**Environmental Measures**	**Decolonization**
Obtain expert consultation from persons with experience in infection control and the epidemiology of MDROS, either in-house or through outside consultation, for assessment of the local MDRO problem and guidance in the design, implementation and evaluation of appropriat4e control measures. *(IB)* Provide necessary leadership, funding and day-to-day oversight to implement interventions selected. *(IB)* Evaluate healthcare system factors for role in creating or perpetuating MDRO transmission, including staffing levels, education and training, availability of consumable and durable resources; communication processes, and adherence to infection control measures.*(IB)* Update healthcare providers and administrators on the progress and effectiveness of the intensified interventions. *(IB)*	Intensify the frequency of educational programs for healthcare personnel, especially for those who work in areas where MDRO rates are not decreasing. Provide individual or unit-specific feedback when available. *(IB)*	Review the role of antimicrobial use in perpetuating the MDRO problem targeted for intensified intervention. Control and improve antimicrobial use as indicated. Antimicrobial agents that may be targeted include vancomycin, third-[d] generation cephalosporins, anti-anaerobic agents for VRE; third generation cephalosporins for ESBLs; and quinolones and carbapenems. *(IB)*	Calculate and analyze incidence rates of target MDROs (single isolates/patient; location-, service-specific) *(IB)* Increase frequency of compiling, monitoring antimicrobial susceptibility summary reports *(II)* Implement laboratory protocols for storing isolates of selected MDROs for molecular typing; perform typing if needed *(IB)* Develop and implement protocols to obtain active surveillance cultures from patients in populations at risk. *(IB)* (See recommendations for appropriate body sites and culturing methods.) Conduct culture surveys to assess efficacy of intensified MDRO control interventions. Conduct serial (e.g., weekly) unit-specific point prevalence culture surveys of the target MDRO to determine if transmission has decreased or ceased.*(IB)* Repeat point-prevalence culture-surveys at routine intervals and at time of patient discharge or transfer until transmission has ceased. *(IB)* If indicated by assessment of the MDRO problem, collect cultures to assess the colonization status of roommates and other patients with substantial exposure to patients with known MDRO infection or colonization. *(IB)* Obtain cultures from HCP for target MDROs when there is epidemiologic evidence implicating the staff member as a source of ongoing transmission. *(IB)*	**Use of Contact Precautions:** **Implement Contact Precautions (CP) routinely for all patients colonized or infected with a target MDRO. *(IA)*** **Don gowns and gloves *before or upon entry* to the patient's room or cubicle. *(IB)*** **In LTCFs, modify CP to allow MDRO-colonized/infected patients whose site of colonization or infection can be appropriately contained and who can observe good hand hygiene practices to enter common areas and participate in group activities** **When active surveillance cultures are obtained as part of an intensified MDRO control program, implement CP until the surveillance culture is reported negative for the target MDRO *(IB)*** No recommendation is made for universal use of gloves and/or gowns. (*Unresolved issue)* **Implement policies for patient admission and placement as needed to prevent transmission of the problem MDRO. *(IB)*** When single-patient rooms are available, assign priority for these rooms to patients with known or suspected MDRO colonization or infection. Give highest priority to those patients who have conditions that may facilitate transmission, e.g., uncontained secretions or excretions. When single-patient rooms are not available, cohort patients with the same MDRO in the same room or patient-care area. *(IB)* When cohorting patients with the same MDRO is not possible, place MDRO patients in rooms with patients who are at low risk for acquisition of MDROs and associated adverse outcomes from infection and are likely to have short lengths of stay. (*II)* **Stop new admissions to the unit or facility if transmission continues despite the implementation of the intensified control measures. *(IB)*** 	Implement patient.-dedicated use of non-critical equipment *(IB)* Intensify and reinforce training of environmental staff who work in areas targeted for intensified MDRO control. Some facilities may choose to assign dedicated staff to targeted patient care areas to enhance consistency of proper environmental cleaning and disinfection services *(IB)* **Monitor cleaning performance to ensure consistent cleaning and disinfection of surfaces in close proximity to the patient and those likely to be touched by the patient and HCWs (e.g., bedrails, carts, bedside commodes, doorknobs, faucet handles) *(IB).*** Obtain environmental cultures (e.g., surfaces, shared equipment) only when epidemiologically implicated in transmission *(IB)* **Vacate units for environmental assessment and intensive cleaning when previous efforts to control environmental transmission have failed *(II)*** 	Consult with experts on a case-by-case basis regarding the appropriate use of decolonization therapy for patients or staff during limited period of time as a component of an intensified MRSA control program *(II)* When decolonization for MRSA is used, perform susceptibility testing for the decolonizing agent against the target organism or the MDRO strain epidemiologically implicated in transmission. Monitor susceptibility to detect emergence of resistance to the decolonizing agent. Consult with microbiologists for appropriate testing for mupirocin resistance, since standards have not been established. Do not use topical mupirocin routinely for MRSA decolonization of patients as a component of MRSA control programs in any healthcare setting. *(IB)* Limit decolonization to HCP found to be colonized with MRSA who have been epidemiologically implicated in ongoing transmission of MRSA to patients. *(IB)* No recommendation can be made for decolonization of patients who carry VRE or MDR-GNB.