APIC Guideline for Selection and Use of Disinfectants

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APIC

ASSOCIATION FOR PROFESSIONALS IN
INFECTION CONTROL AND EPIDEMIOLOGY, INC.

SUPPLEMENT

APIC GUIDELINES FOR INFECTION CONTROL PRACTICE

The Association for Professionals in Infection Control and Epidemiology, Inc. (APIC) Board of Directors and Guidelines Committee are pleased to present the "APIC Guideline for Selection and Use of Disinfectants."

William A. Rutala, PhD, MPH, CIC, was selected to revise the previously published "APIC Guideline for Selection and Use of Disinfectants" because of his recognized expertise in infection control and extensive research with disinfectants. Initial drafts were reviewed by the APIC Guidelines Committee, key individuals, and professional organizations before the publication of the draft document in the August 1995 issue of AJIC, soliciting further comments. All written comments were reviewed by the APIC Guidelines Committee and revisions were made. The Guideline was finalized by the Committee in February 1996 and approved by the APIC Board of Directors in March 1996.

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APIC guideline for selection and use of disinfectants*

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The need for appropriate disinfection and sterilization has been emphasized by numerous articles documenting infection after improper reprocessing of patient care items. Because it is unnecessary to sterilize all patient care items, hospital policies must identify whether disinfection or sterilization is indicated on the basis of each item's intended use. In 1982 the Centers for Disease Control and Prevention (CDC) prepared a "Guideline for Hospital Environmental Control," which provided specific directions for the selection and use of disinfectants.[1] A revised version of

this guideline, entitled "Guideline for Handwashing and Hospital Environmental Control, 1985," was published in November 1985.[2] This latter guideline did not recommend chemical germicides that were formulated for use on medical equipment or environmental surfaces in health care facilities. Rather, the revised CDC guideline focused on strategies for disinfection and sterilization of medical (equipment used in the health care setting.

The purpose of this revised Association for Professionals in Infection Control and Epidemiology, Inc. (APIC) Guideline, which is an updated version of previous publications,[3-6] is to assist health care professi~onals in their decisions involving the judicious selection and proper use of specific disinfectants. In the preparation of this guideline, articles in the scientific literature were used to augment the manufacturers' label claims because these claims were not consistently verifiables[7] Disinfectant failures noted at variance to label claims may be caused by deficiencies in testing methods[7] or by improperly conducted tests[8] In addition, in-use testing has not been incorporated into all Environmental Protection Agency (EPA) methods (e.g., Association of Official Analytical Chemists [AOAC] tuberculocidal activity test), and fa:ilures have been demonstrated when some disinfectants are subjected to conditions, such as dilution, age, and presence of organic matter, that challenge their antimicrobial activity.[9] It should also be recognized that EPA registration claims are based on microbicidal efficacy data submitted by manufacturers. The EPA does not independently test disinfectants before their registration, but in 1990 the EPA resumed postregistration testing of chemical sterilants to ensure that they satisfy their registered label claims.

DEFINITIONS

For the purpose of this guideline, the following definitions will be used:

Sterilization is the complete elimination or destruction of all forms of microbial life. It is accomplished by either physical or chemical processes. Steam under pressure, dry heat, low temperature sterilization processes (ethylene oxide [ETO] gas, plasma sterilization) and liquid chemicals are the principal sterilizing agents used. The term *sterilization is* intended to convey an absolute meaning, not a relative one.

Disinfection describes a process that eliminates

BACTERIALSPORES
Bacillus   subtilis
⇓
MYCOBACTERIA
Mycobacterium   tuberculosis
⇓
NONLIPID OR SMALLVIRUSES
poliovirus
↓
FUNGI
Trichophyton spp.

VEGETATIVE BACTERIA
Pseudomonas   aeruginosa
Staphylococcus   aureus
⇓
LIPID OR MEDIUM-SIZED VIRUSES
herpes simplex virus
hepatitis B virus
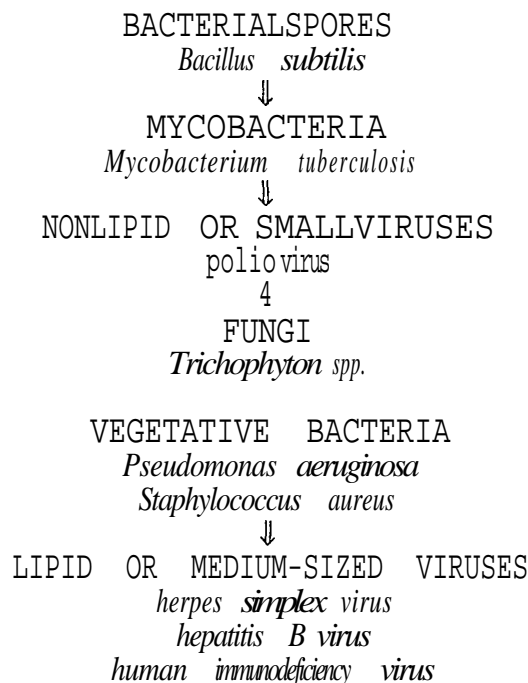*human   immunodeficiency   virus*

Fig. 1. Descending order of resistance to germicidal chemicals. This hierarchy considers broad classifications of microbial categories. It is considered a rough guide to general susceptibility of microorganisms to disinfectants. Adapted from Favero MS, Bond WV Chemical disinfection of medical and surgical materials. In: Block SS, ed. Disinfection, sterilization and preservation. 4th ed. Philadelphia: Lea & Febiger, 1991:621.

many or all pathogenic microorganisms, with the exception of bacterial spores, from inanimate objects. In health care settings, this is generally accomplished by the use of liquid chemicals or wet pasteurization. The efficacy of disinfection is affected by a number of factors, each of which may nullify or limit the efficacy of the process. Some of the factors that have been shown to affect disinfection efficacy are the previous cleaning of the object, the organic load on the object, the type (Fig. 1) and level of microbial contamination, the concentration of and exposure time to the germicide, the physical configuration of the object (e.g., crevices, hinges, lumens), and the temperature and pH of the disinfection process. More extensive consideration of these and other factors that affect both disinfection and sterilization may be found in several references. 3, [6, 10-13] Chemical disinfectants can be classified by several schemes. This guideline uses the terminology used by the CDC's "Guideline for Handwashing and Hospital

Table 1. Classification of devices, processes, and germicidal products

Device classification	Devices (examples)	Spaulding process classification	EPA product classification
Critical (enters sterile tissue or vascular system)	Implants, scalpels, needles, other surgical instruments, etc.	*Sterilization*—sporicidal chemical; prolonged contact	Sterilant/disinfectant
Semicritical (touches mucous membranes [except dental])	Flexible endoscopes, laryngoscopes, endotracheal tubes, and other similar instruments	*High-level disinfection*—sporicidal chemical; short contact	Sterilant/disinfectant
	Thermometers, hydrotherapy tanks	*Intermediate-level disinfection*	Hospital disinfectant with label claim for tuberculocidal activity
Noncritical (touches intact skin)	Stethoscopes, tabletops, bedpans, etc.	*Low-level disinfection*	Hospital disinfectant without label claim for tuberculocidal activity

Modified from Favero MS, Bond WW. Chemical disinfection of medical and surgical materials. In: Block SS, ed. Disinfection, sterilization and preservation. 4th ed. Philadelphia: Lea & Febiger 1991:627.

ronmental Control, 1985,"[2] in which the levels of disinfection are defined as *sterilization, high-level disinfection, intermediate-level disinfection,* and *low-level disinfection.* These terms were also used in the CDC's "Guidelines for the Prevention of Transmission of Human Immunodeficiency Virus and Hepatitis B Virus to Health-Care and Public-Safety Workers."[14]

High-level disinfection can be expected to destroy all microorganisms, with the exception of high numbers of bacterial spores. *Intermediate-level disinfection* inactivates Mycobacterium tuberculosis, vegetative bacteria, most viruses, and most fungi, but it does not necessarily kill bacterial spores. *Low-level disinfection* can kill most bacteria, some viruses, and some fungi, but it cannot be relied on to kill resistant microorganisms such as tubercle bacilli or bacterial spores.

Cleaning is the removal of all foreign material (e.g., soil, organic material) from objects. It is normally accomplished with water, mechanical action, and detergents or enzymatic products. Failure to remove foreign matter (e.g., lubricants, soils) from an object before a disinfection or sterilization process is likely to render the process ineffective.[15-18] Meticulous physical cleaning must precede disinfection and sterilization procedures. Studies have shown that manual and mechanical cleaning of endoscopes achieves approximately a 4 log reduction of contaminating organisms.[15] Thus cleaning alone is very effective in reducing the number of microorganisms present on contaminated equipment. A *germicide* is an agent that destroys microorganisms, particularly pathogenic organisms ("germs"). Other agents designated by

words with the suffix *-cide* (e.g., virucide, fungicide, bactericide, sporicide, tuberculocide) destroy the microorganisms identified by the prefix. For example, a *bactericide* is an agent that kills bacteria.[1, 10, 11, 19] Chemicals used for the purpose of destroying all forms of microbial life, including fungal and bacterial spores, are called *chemical sterilants.* These same chemical sterilants may also be part of the high-level disinfection process when used for shorter exposure periods. A *disinfectant* is a germicide that inactivates virtually all recognized pathogenic microorganisms but not necessarily all microbial forms (e.g., bacterial endospores) on inanimate objects. As of June 1993, the Food and Drug Administration (FDA) has primary responsibility for the premarket review of safety and efficacy requirements for liquid chemical germicides that are sterilants intended for use on critical and semicritical devices. The EPA has primary responsibility for premarket review of general-purpose disinfectants used on noncritical items.[20] An *antiseptic* is a chemical germicide formulated for use on skin or tissue and should not be used to decontaminate inanimate objects. The selection and use of antiseptics are extensively discussed in another publication.[21] Antiseptics are registered and regulated by the FDA.

A RATIONAL APPROACH TO DISINFECTION AND STERILIZATION

In 1968 a rational approach to disinfection and sterilization of patient care items or equipment was devised by E. H. Spaulding.[11] This classification scheme is so clear and logical that it has been

retained, refined, and successfully used by infection control professionals (ICPs) and others when planning methods for disinfection or sterilization.[1-4, 10] Spaulding[11] believed that the nature of disinfection could be understood more readily if instruments and items for patient care were divided into three categories according to degree of risk of infection involved in the use of the items. The three categories of items he described were as follows: *critical*, *semicritical*, and *noncritical*. Table 1 correlates the three device classifications (critical, semicritical, and noncritical) with Spaulding's process classification and the EPA's product classifications.

Critical items

Items assigned to the *critical* category present a high risk of infection if contaminated with any microorganism, including bacterial spores. It is critical that objects entering sterile tissue or the vascular system be kept sterile. This category includes surgical instruments, cardiac and urinary catheters, implants, and needles. Most of the items in this category should be purchased as sterile or should be sterilized by steam under pressure if possible. If heat labile, the object may be treated with ETO or other low temperature sterilization processes. Table 2 lists several germicides categorized as chemical sterilants. These include 2% glutaraldehyde-based formulations, 6% stabilized hydrogen peroxide, and peracetic acid. Chemical sterilants can be relied on to produce sterility only if adequate cleaning precedes treatment and if proper guidelines with regard to organic load, contact time, temperature, and pH are met.

Semicritical items

Semicritical items are those objects that come in contact with mucous membranes or skin that is not intact. These items must be free of all microorganisms, with the exception of high numbers of bacterial spores. Intact mucous membranes are generally resistant to infection by common bacterial spores but are susceptible to other organisms, such as tubercle bacilli and viruses. Respiratory therapy and anesthesia equipment, endoscopes, and cervical diaphragm fitting rings are included in this category. Semicritical items generally require high-level disinfection with wet pasteurization or chemical disinfectant. Glutaraldehyde, stabilized hydrogen peroxide, chlorine, and peracetic acid are dependable high-level disinfectants, provided the factors influencing germicidal

procedures are considered (Table 2). Heat sterilization is the preferred method of between-patient processing of heat-stable medical instruments because it provides the widest margin of safety, even though high-level disinfection with a liquid chemical disinfectant would provide a patient-safe device. When selecting a disinfectant for use with certain patient care items, the chemical compatibility after extended use with the items must also be considered. For example, although chlorine is considered a high-level disinfectant, it is generally not used for disinfecting semicritical items because of its corrosive effects.

It is recommended that semicritical items be rinsed with sterile water after disinfection to prevent contamination with organisms that may be present in tap water, such as nontuberculous mycobacteria and *Legionella*.[2, 5, 22-26] In circumstances under which a sterile water rinse is not feasible, a tap water rinse should be followed by an alcohol rinse and forced-air drying.[22, 24, 27, 28] Introduction of forced-air drying significantly reduces bacterial contamination of stored endoscopes, presumably by removing the wet environment favorable for bacterial growth.[18, 27]

Some semicritical items (e.g., hydrotherapy tanks used for patients whose skin is not intact, thermometers) may require only intermediate-level disinfection. Intermediate-level disinfectants (e.g., chlorine, phenolics, iodophor) inactivate *M. tuberculosis*, vegetative bacteria, most viruses, and most fungi but do not necessarily kill bacterial spores.

Noncritical items

Noncritical items come in contact with intact skin but not with mucous membranes. Intact skin acts as an effective barrier to most microorganisms, and sterility is not critical. Examples of noncritical items include bedpans, blood pressure cuffs, crutches, bed rails, linens, some food utensils, bedside tables, and patient furniture. Most noncritical reusable items may be disinfected where they are used and do not need to be transported to a central processing area. There is generally little risk of transmitting infectious agents to patients by means of noncritical items[29]; however, these items could potentially contribute to secondary transmission by contaminating hands of health care workers or by contact with medical equipment that subsequently comes in contact with patients.[10, 30] The low-level disinfectants listed in Table 2 may be used for noncritical items.

Table 2. Methods of sterilization and disinfection

Object	Sterilization		Disinfection		
	Critical items (will enter tissue or vascular system or blood will flow through them)		High-level (semicritical items [except dental[a]] will come in contact with mucous membrane or nonintact skin)	Intermediate-level (some semicritical items[b] and noncritical items)	Low-level (noncritical items; will come in contact with intact skin)
	Procedure	Exposure time (hr.)	Procedure (exposure time ≥ 20 min.)[c, d]	Procedure (exposure time ≤ 10 min.)	Procedure (exposure time ≤ 10 min.)
Smooth hard surface[b]	A	MR	C	G[e]	H
	B	MR	D	H	I
	C	MR	E	J	J
	D	6	F[f]	K	K
	E	MR	G		L
Rubber tubing and catheters[d]	A	MR	C		
	B	MR	D		
	C	MR	E		
	D	6	F[f]		
	E	MR			
Polyethylene tubing and catheters[d, g]	A	MR	C		
	B	MR	D		
	C	MR	E		
	D	6	F[f]		
	E	MR			
Lensed instruments	B	MR	C		
	C	MR	D		
	D	6	E		
	E	MR			
Thermometers (oral and rectal)[h]				H[h]	
Hinged instruments	A	MR	C		
	B	MR	D		
	C	MR	E		
	D	6			
	E	MR			

Adapted from Simmons BP. Guideline for hospital environmental control. Am J Infect Control 1983;11:97-115.

A, Heat sterilization, including steam or hot air (see manufacturer's recommendations).

B, Ethylene oxide gas (see manufacturer's recommendations).

C, Glutaraldehyde-based formulations (2%). (Caution should be exercised with all glutaraldehyde formulations when further in-use dilution is anticipated.).

D, Stabilized hydrogen peroxide 6% (will corrode copper, zinc, and brass).

E, Peracetic acid, concentration variable but ≤1% is sporicidal.

F, Wet pasteurization at 70° C for 30 minutes after detergent cleaning.

G, Sodium hypochlorite (5.2% household bleach) 1:50 dilution (1000 ppm free chlorine).

H, Ethyl or isopropyl alcohol (70% to 90%).

I, Sodium hypochlorite (5.2% household bleach) 1:500 dilution (100 ppm free chlorine).

J, Phenolic germicidal detergent solution (follow product label for use-dilution).

K, Iodophor germicidal detergent solution (follow product label for use-dilution).

L, Quaternary ammonium germicidal detergent solution (follow product label for use-dilution).

MR, Manufacturer's recommendations.

[a]Semicritical dental items (e.g. handpieces, amalgam condensers) should be heat sterilized; refer to text for details.

[b]See text for discussion of hydrotherapy.

[c]The longer the exposure to a disinfectant, the more likely it is that all microorganisms will be eliminated. Ten minutes' 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 minutes' exposure is the minimum time needed to reliably kill *M. tuberculosis* and nontuberculous mycobacteria with glutaraldehyde.

[d]Tubing must be completely filled for chemical disinfection; care must be taken to avoid entrapment of air bubbles during immersion.

[e]Used in laboratory where cultures or concentrated preparations or microorganisms have spilled. This solution may destroy some surfaces.

[f]Pasteurization (washer disinfector) of respiratory therapy and anesthesia equipment is a recognized alternative to high-level disinfection. Some data challenge the efficacy of some pasteurization units (J Hosp Infect 1983;4:119-208).

[g]Thermostability should be investigated when appropriate.

[h]Do not mix rectal and oral thermometers at any stage of handling or processing.

CHANGES SINCE 1981

As a guide to the appropriate selection and use of disinfectants, a table was prepared by the CDC in 1981 and is presented here in modified form (Table 2). This current table contains several changes from the original CDC guideline[1] and one change from the 1990 APIC Guideline.[5] First, formaldehyde-alcohol has been deleted as a chemical sterilant and high-level disinfectant because, with the exception of dialysis equipment, it no longer has a role in disinfection strategies. It is corrosive, irritating, toxic, and not commonly used.[31, 32] Second, the chemical sterilant demand-release chlorine dioxide[33-35] is deleted from the table because it is no longer commercially available, and peracetic acid[36, 37] has been added to the table. Third, 3% phenolic and iodophors have been deleted as high-level disinfectants because of their unproven efficacy against bacterial endospores, *M. tuberculosis*, and some fungi.[34] Fourth, isopropyl and ethyl alcohols have been excluded as high-level disinfectants because of their inability to inactivate bacterial spores and because of the inability of isopropyl alcohol to inactivate hydrophilic viruses.[38] Fifth, a 1:16 dilution of 2.0% glutaraldehyde–7.05% phenol–1.2% sodium phenate (which contains 0.13% glutaraldehyde, 0.44% phenol, and 0.075% sodium phenate when diluted) has been deleted as a high-level disinfectant because of numerous scientific publications that demonstrate a lack of bactericidal activity in the presence of organic matter; a lack of fungicidal, tuberculocidal, and sporicidal activity; and reduced virucidal activity.[34, 39-47] This product and another diluted glutaraldehyde were removed from the marketplace by the EPA, FDA, and Federal Trade Commission in 1991. Sixth, the exposure time required to achieve high-level disinfection has been changed from a period of 10 to 30 minutes to a period of 20 minutes or more.[15, 39, 41, 42, 48-51]

PROBLEMS WITH DISINFECTION AND STERILIZATION OF HEALTH CARE EQUIPMENT

Concerns with Spaulding scheme

One problem associated with the Spaulding scheme is that of oversimplification. For example, the system does not consider problems with processing complicated medical equipment, which is often heat labile, or problems of inactivating certain microorganisms. In some situations, it is therefore still difficult to choose a level of disinfection after considering the categories of risk to patients. This is especially true for a few medical devices (e.g., arthroscopes, laparoscopes) in the critical category because there is a controversy regarding whether we should sterilize or high-level disinfect these patient care items.[22,52] Sterilization would not be a problem if these items could be steam sterilized, but most of these items are heat labile, and sterilization is achieved by using ETO, which may be too time-consuming for routine use between patients. Whereas new technology is making it easier to sterilize these items, evidence that sterilization of these items improves patient care by reducing the infection risk is lacking.[53-55] Presumably these reasons account for the fact that many procedures done with arthroscopes and laparoscopes are performed with equipment that has been processed by high-level disinfection, not sterilization.[22, 53] Ideally, biopsy forceps or other cutting instruments that break the mucosal barrier and laparoscopes, arthroscopes, and other scopes that enter normally sterile tissue should be subjected to a sterilization process before each use.[23]

This is also true for equipment in the semicritical category such as flexible endoscopes, which may be heat labile and with which there may be difficulty in exposing organisms to a sterilization process. For example, is the endoscope used for upper gastrointestinal tract examination still a semicritical item when it is used with sterile biopsy forceps or when it is used in a patient who is bleeding heavily from esophageal varices? Provided that high-level disinfection is achieved and all microorganisms with the exception of a high number of bacterial spores have been removed from the endoscope, then the endoscope should not represent an infection risk and should remain in the semicritical category.[56]

Several other problems are associated with the disinfection of patient care items.[6] The optimal contact times and disinfection schemes are not known for all equipment. For this reason, disinfectant strategies for several semicritical items (e.g., endoscopes, applanation tonometers, cryosurgical instruments, diaphragm fitting rings) are highly variable and are discussed further in this guideline. Although additional studies are needed to determine whether simplified disinfecting procedures are efficacious in a clinical setting, it is prudent to follow the CDC and the APIC guidelines until studies have defined effective alternative processes.[2, 5, 57, 58]

Endoscopes

High-level disinfection can be expected to destroy all microorganisms, with the exception of

high numbers of bacterial spores. An immersion time of ≥ 20 minutes in 2% glutaraldehyde is required to adequately disinfect semicritical items such as endoscopes between patient procedures, particularly in view of the disputed tuberculocidal efficacy of glutaraldehyde-based disinfectants.[15, 39, 41, 42, 48-51] Flexible endoscopic instruments are particularly difficult to disinfect and easy to damage because of their intricate design and delicate materials. It must be highlighted that meticulous cleaning must precede any sterilization or disinfection procedures or outbreaks of infection may occur.

Examining reports of nosocomial infections related only to endoscopes, one finds that 281 infections were transmitted by gastrointestinal endoscopy and 96 were transmitted by bronchoscopy. The clinical spectrum of these infections ranged from asymptomatic colonization to death. *Salmonella* species and *Pseudomonas aeruginosa* were repeatedly 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. Major reasons for transmission were inadequate cleaning, improper selection of a disinfecting agent, or failure to follow recommended cleaning and disinfection procedures.[59] One multistate investigation found that 23.9% of the bacterial cultures from the internal channels of 71 gastrointestinal endoscopes grew 100,000 colonies or more of bacteria after completion of all disinfection or sterilization procedures and before use in the next patient.[60] Automatic endoscope reprocessing machines have also been linked to outbreaks of infection[61] or colonization.[62] Outbreaks involving endoscopic accessories,[63, 64] such as suction valves and biopsy forceps, support a recommendation that if such an item cannot be cleaned of all foreign matter, it should be steam sterilized, when heat stable.[65]

Clearly, there is a need for further development and redesign of automated endoscope reprocessing machines[66] and endoscopes[67] so they do not represent a potential source of infectious agents. A redesigned endoscope was introduced that includes a reusable endoscope without channels and a sterile sheath set comprising a single disposable unit: a sheath; air, water, and suction channels; a distal window; and a cover for the endoscope control body. All contaminated surfaces, including the channels, are then discarded, thereby eliminating any concern for cross-transmission of infectious agents from the previous patients. Fur-

ther clinical trials and microbiologic evaluations are needed to document the comparability, cost-effectiveness, safety, and reduced infection risk of this system.

Recommendations for the cleaning and disinfection of endoscopic equipment have been published and should be followed.[23, 24, 68, 69] In general, endoscope disinfection involves six steps, which are as follows: (1) clean—mechanically clean external surfaces, ports, and internal channels with water and a detergent or enzymatic detergent; (2) rinse and drain channels; (3) disinfect—immerse endoscope in high-level disinfectant and perfuse disinfectant into the suction/biopsy channel and air and water channels and expose for at least 20 minutes; (4) rinse—the endoscope and channels should be rinsed with sterile water; if this is not feasible use tap water followed with an alcohol rinse; (5) dry—the insertion tube and inner channels should be dried by means of forced air after disinfection and before storage; and (6) store—the endoscope should be stored in a way that prevents recontamination.

FDA labeling requirements

As mentioned, the FDA now regulates the efficacy claims for chemical sterilants. All chemical sterilants (e.g., glutaraldehyde-based solutions) that are used for sterilization or high-level disinfection and come in contact with medical devices require premarket clearance from the FDA (called 510[K] -named after that section of the Food, Drug and Cosmetic Act describing the process). In April 1994 a chemical sterilant manufacturer received the first 510(K) clearance for its glutaraldehyde-based solutions from the FDA. The time and temperature specified for one formulation of 2.4% alkaline glutaraldehyde with a high-level disinfection claim (100% kill of *M. tuberculosis*) was 45 minutes at 25° C (77° F). One would expect similar competitive 2% alkaline glutaraldehyde products to have comparable label claims. Additionally, the FDA requires that the manufacturers provide additional use instructions to the health care worker.

The data required by the FDA are quite rigorous, requiring the quantitative tuberculocidal test and 100% kill of *M. tuberculosis* for high-level disinfectant claims. Because the quantitative test does not allow for cleaning, is conducted in the presence of 2% horse serum (a protein load), and uses an extremely high number of organisms (100,000 to 1,000,000), it is necessary to have an extended immersion time (e.g., 45 minutes) and elevated

temperature (25° C) to inactivate 100% of the mycobacteria. Several investigators, however, have shown that cleaning alone of endoscopic equipment is extremely effective in eliminating microbial contaminants. These studies have shown a mean 4 log (99.99%) reduction in microbial contaminants with cleaning alone.[70-73] Cleaning is a very effective adjuvant because it removes pathogenic microorganisms on inanimate objects and also removes organic matter that may interfere with the microbicidal activity of the germicide. Because neither the manufacturers nor the FDA has control over the cleaning techniques, a specific label statement cannot be made with respect to the potential decrease in immersion time. In the absence of cleaning and the presence of proteinaceous materials with high microbial loads, immersion in a 2.4% alkaline glutaraldehyde for 45 minutes at 25° C may be necessary for 100% tuberculocidal kill. This statement should not be interpreted to mean that prolonged immersion is an adequate substitute for proper cleaning before high-level disinfection or sterilization.

When proper cleaning is used, multiple studies demonstrate that *M. tuberculosis* is effectively destroyed by a 20-minute immersion time[15, 39, 41, 42, 48-51] in glutaraldehyde and other chemical sterilants at 20° C. The "APIC Guideline for Infection Prevention and Control in Flexible Endoscopy" recommendation of 20 minutes or longer at 20° C for high-level disinfection presumes precleaning with an enzymatic detergent[74] or detergent that removes debris and significantly reduces microbial contaminants.

Laparoscopes and arthroscopes

Although high-level disinfection appears to be the minimum standard for processing laparoscopes and arthroscopes between patients,[22, 52, 53, 75] there continues to be debate regarding this practice.[55, 76] Proponents of high-level disinfection refer to membership surveys[53] or institutional experiences[54] involving more than 117,000 and 10,000 laparoscopic procedures, respectively, that cite a low risk of infection (< 0.3%) when high-level disinfection is used for gynecologic laparoscopic equipment. Only one infection in the membership survey series was believed to be related to spores. In addition, studies conducted by Corson et al.[77, 78] demonstrated growth of common skin microorganisms (e.g., *Staphylococcus epidermidis*, diphtheroids) from the umbilical area even after skin preparation with povidone-iodine and ethyl alcohol. Similar organisms were recovered in some cases from the pelvic serosal surfaces and from the laparoscopic telescopes, suggesting

that the microorganisms were probably carried from the skin into the peritoneal cavity. 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; these include the following: limited number of organisms (usually < 10) introduced into the peritoneal cavity, minimal damage to inner abdominal structures with little devitalized tissue, tolerance of the peritoneal cavity to small numbers of spore-forming bacteria, simplicity of cleaning and disinfection of equipment, relative nature of surgical sterility, and lack of epidemiologic evidence that high-level disinfection increases the infection risk.[54]

As with laparoscopes and other equipment that enters sterile body sites, arthroscopes ideally should be sterilized before use. In the United States, however, they commonly undergo high-level disinfection.[22, 53] Presumably this is because the incidence of infection is low and the few infections that occur are probably unrelated to the use of high-level disinfection rather than sterilization. In a retrospective study of 12,505 arthroscopic procedures, Johnson and associates[55] found an infection rate of 0.04% (five infections) when arthroscopes were soaked in 2% glutaraldehyde for 15 to 20 minutes. Interestingly, four infections were caused by *Staphylococcus aureus*, and the other was an anaerobic streptococcal infection. Because these organisms are very susceptible to 2% glutaraldehyde, the source of these infections was probably the patient's skin. Although only limited data are available, there is no evidence to demonstrate that high-level disinfection of arthroscopes poses an infection risk to the patient. Although the debate regarding high-level disinfection versus sterilization of laparoscopes and arthroscopes will go unsettled until there are well-designed, randomized clinical trials, the CDC and APIC guidelines are appropriate.[2, 5] That is, laparoscopes, arthroscopes, and other scopes that enter normally sterile tissue should be subjected to a sterilization procedure before each use; if this is not feasible, they should receive at least high-level disinfection. If high-level disinfection is used, a sterile water rinse is required to prevent contamination with tap water organisms. After rinsing, the scopes must be dried according to a method that does not recontaminate the item.

Tonometers, diaphragm fitting rings, cryosurgical instruments

Disinfection strategies for other semicritical items (e.g., applanation tonometers, cryosurgical

instruments, and diaphragm fitting rings) are highly variable. For example, one study revealed that no uniform technique was in use for disinfection of applanation tonometers, with disinfectant contact times varying from less than 15 seconds to 20 minutes.[22] Concern regarding transmission of viruses (e.g., herpes simplex virus [HSV], adenovirus 8, HIV) by tonometer tips has prompted CDC disinfection recommendations.[57] These recommendations are that the instrument be wiped clean and disinfected for 5 to 10 minutes with either 3% hydrogen peroxide, 500 parts per million (ppm) chlorine, 70% ethyl alcohol, or 70% isopropyl alcohol. After disinfection, the device should be thoroughly rinsed in tap water and dried before use. Although these disinfectants and exposure times should kill microorganisms of relevance in ophthalmology, each of these disinfectants has not been tested against all relevant pathogens.[79] The American Academy of Ophthalmology also has developed specific guidelines for preventing infection in ophthalmology practice, but they only consider certain infectious agents (e.g., HIV, herpes, adenovirus).[80] Because a short and simple cleaning procedure is desirable in the clinical setting, swabbing the tonometer tip with a 70% isopropyl alcohol wipe is sometimes practiced.[79] Preliminary reports suggest that wiping the tonometer tip with an alcohol swab and then allowing the alcohol to evaporate may be an effective means of eliminating HSV-1, HIV-1, and adenovirus 8.[79, 81, 82] 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 studies 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.[83, 84] Therefore it is recommended that the tonometer be immersed in the germicides listed here for at least 5 minutes.

No studies have evaluated disinfection techniques for other items that contact mucous membranes, such as diaphragm fitting rings, cryosurgical probes, or vaginal probes used in sonographic scanning. Lettau et al.[58] of the CDC supported a diaphragm fitting ring manufacturer's recommendation, which involved a soap-and-water wash followed by a 15-minute, 70% alcohol immersion. This disinfection method should be adequate to inactivate HIV-1, hepatitis B virus (HBV), and HSV, even though alcohols are not classified as high-level disinfectants because their activity against picornaviruses is somewhat limited.[38] There are no data on the inactivation of human papillomavirus by alcohol or other disinfectants because in vitro replication of complete virions has not been achieved. Thus, although isopropyl alcohol for 15 minutes should kill microorganisms of relevance in gynecology, there are no clinical studies that provide direct support for this procedure. Cryosurgical probes should be high-level disinfected. A condom may be used to cover the vaginal probe used in sonographic scanning. A new condom should be used to cover the probe with each new patient; because condoms may fail, however, high-level disinfection of the probe is necessary after each use.

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 disease transmission.[85, 86] The American Dental Association recommends that surgical and other instruments that normally penetrate soft tissue or bone (e.g., forceps, scalpels, bone chisels, scalers, and surgical burs) are classified as critical and must be sterilized or discarded after each use. Instruments that are not intended to penetrate oral soft tissues or bone (e.g., amalgam condensers, air/water syringes) but may come in contact with oral tissues are classified as semicritical and should also be sterilized after each use.[87] This is consistent with the recommendations from the CDC and the FDA.[88, 89] Handpieces that cannot be heat sterilized should be retrofitted to attain heat tolerance. Handpieces that cannot be retrofitted and thus cannot be heat sterilized should not be used.[89] Chemical disinfection is not recommended for critical or semicritical dental instruments that can be heat sterilized. Methods of sterilization that may be used for critical and semicritical dental instruments and materials that are heat stable include the following: steam under pressure (autoclave), heat/chemical vapor, and dry heat, following manufacturers' recommendations. ETO may not be an effective means of sterilization because it may be difficult to ensure that the internal portions of the handpieces are adequately cleaned and dried before ETO processing. Consideration must be given to the effect that a sterilization process may have on instruments and materials.

Uncovered operatory surfaces (e.g., countertops, chair switches, light handles) should be disinfected between patients. This can be accomplished by use of a disinfectant that is registered with the EPA as a "hospital disinfectant." There are several categories of such products.[87, 90, 91] If

Table 3. Inactivation of HBV and HIV by disinfectants

Disinfectant	Concentration inactivating 10⁶ HBV in ST, 10 min., 20° C*	Concentration inactivating 10⁵ HIV in ST, ≤10 min., 25° C†
Ethyl alcohol	ND	50%
Glutaraldehyde	2%	ND
Glutaraldehyde-phenate	0.13% glutaraldehyde—0.44% phenol	ND
Hydrogen peroxide	ND	0.3%
Iodophor	80 ppm	ND
Isopropyl alcohol	70%	35%
Paraformaldehyde	ND	0.5%
Phenolic	ND	0.5%
Sodium hypochlorite	500 ppm	50 ppm

ST, Suspension test; *ND*, no data.
*Data from Bond et al.[92]
†Data from Martin et al.[95] Also see Sattar and Springthorpe[99] for data concerning activity of other disinfectants against HIV.

waterproof surface covers are used to prevent contamination of surfaces and are carefully removed and replaced between patients, the protected surfaces do not need to be disinfected between patients but must be disinfected at the end of the day.

Disinfection of devices contaminated with HBV, HIV, or *M. tuberculosis*

Should we sterilize or high-level disinfect semicritical medical devices contaminated by blood from patients infected with HIV or HBV or by respiratory secretions from a patient with pulmonary tuberculosis? The CDC recommendation for high-level disinfection is appropriate because experiments have demonstrated the effectiveness of high-level disinfectants to inactivate these and other pathogens that may contaminate semicritical devices (Table 3).* Nonetheless, some hospitals modify their disinfection procedures when the endoscopes have been used with a patient known or suspected to be infected with HIV, HBV, or *M. tuberculosis*.[22, 104] This practice is inconsistent with the concept of universal precautions, which presumes that all patients are potentially infected with blood-borne pathogens.[97] Several studies have highlighted the inability to distinguish HIV- or HBV-infected patients from noninfected patients on clinical grounds.[105-107] It is also likely that in many patients mycobacterial infection is not immediately clinically apparent. It should be noted that in most cases hospitals gas-sterilized endoscopic instruments because they believed that this practice reduced the risk of infection.[22, 104] 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 infected with *M. tuberculosis*, HIV, or HBV.

Inactivation of *Clostridium difficile*

Some investigators have also recommended the use of dilute solutions of hypochlorite for routine environmental disinfection of rooms of patients with *C. difficile*–associated diarrhea or colitis.[108] This practice would appear unnecessary because studies have shown that patients without symptoms constitute an important reservoir within the hospital and that person-to-person transmission is the principal means of transmission between patients. Handwashing, barrier precautions, and meticulous environmental cleaning may therefore be equally effective in preventing the spread of *C. difficile*.[109]

Contaminated endoscopes such as colonoscopes can serve as vehicles of transmission. For this reason, investigators have studied commonly used disinfectants and exposure times to assess whether current practices may be placing patients at risk. Data demonstrate that 2% glutaraldehyde reliably kills *C. difficile* spores with short exposure times (≤20 minutes).[46, 110, 111]

Inactivation of Creutzfeldt-Jakob disease (CJD) agent

The only infectious agent that requires unique decontamination recommendations is the prion CJD.[112] CJD is a degenerative neurologic disorder with an incidence rate of one new case in 1 million people per year.[113] Infectivity is tissue dependent with the brain, spinal cord, and eye suspected to have the highest infectivity.[114] It has been transmitted iatrogenically by means of implanted brain

*References 33, 39, 48, 70-72, and 92-103.

electrodes that were disinfected with ethanol and formaldehyde after use on a patient known to have CJD. Iatrogenic transmission has been observed in recipients of contaminated human growth hormone, gonadotropin, and corneal, pericardial and dura mater grafts.[113, 115] The need for special recommendations is due to an extremely resistant subpopulation of prions[116] and the protection afforded this tissue-associated virus. Although discrepancies exist between different studies, they all agree that these prions resist normal inactivation methods. Steam sterilization for at least 30 minutes at a temperature of 132° C (121° C ineffective) in a gravity displacement sterilizer has been recommended as the preferred method for the treatment of contaminated material. When a prevacuum sterilizer is used, 18 minutes at 134° to 138° C has been found to be effective. Immersion in 1 N sodium hydroxide (which is caustic) for 1 hour at room temperature followed by steam sterilization at 121° C for 30 minutes is an alternative procedure for critical and semicritical items.[117-122] Because noncritical patient care items or surfaces (e.g., autopsy tables, floors) have not been involved in disease transmission,[29] these surfaces may be disinfected with either bleach (undiluted, or up to 1:10 dilution) or 1 N sodium hydroxide at room temperature for 15 minutes or less.[117] A formalin–formic acid procedure is required for inactivating virus infectivity in tissue samples from patients with CJD.[123]

OSHA blood-borne pathogen standard

In December 1991, the Occupational Safety and Health Administration (OSHA) promulgated a standard entitled "Occupational Exposure to Bloodborne Pathogens; Final Rule" to eliminate or minimize occupational exposure to blood-borne pathogens.[124] One component of this requirement is that all equipment, environmental surfaces, and working surfaces should be cleaned and decontaminated with an appropriate disinfectant after contact with blood or other potentially infectious materials. Although the OSHA standard does not specify the type of disinfectant or procedure, the OSHA compliance document[125] suggests that a germicide must be tuberculocidal to kill HBV. The document thus suggests that a tuberculocidal agent should be used to clean blood spills on noncritical surfaces. This recommendation is inconsistent with data that demonstrate that non-tuberculocidal quaternary ammonium compounds inactivate HBV.[103] Nonetheless, to follow the OSHA compliance document a tuberculocidal

disinfectant (e.g., phenolic, chlorine) would be needed to clean a blood spill. This has caused concern among housekeeping managers, who try to find disinfectant detergents claiming to be tuberculocidal on the assumption that such products would be effective in eliminating transmission of HBV. This directive can be questioned on a practical level for three reasons. First, nontuberculocidal disinfectants such as quaternary ammonium compounds inactivate HBV.[103] Second, noncritical surfaces are rarely involved in disease transmission.[29] Third, the exposure times that manufacturers use to achieve their label claims are not used in health care settings to disinfect noncritical surfaces. For example, to make a label claim against HIV, HBV, or *M. tuberculosis*, a manufacturer must demonstrate inactivation of these organisms when exposed to a disinfectant for 10 minutes. This exposure cannot be practically achieved in a health care setting without immersion. Alternatively, a hospital could use the scientific literature and use any EPA-registered hospital disinfectant (e.g., phenolic, chlorine, quaternary ammonium compounds) for cleaning blood spills on noncritical surfaces. However, this practice could result in an OSHA citation for noncompliance with the rule.

Toxicologic and environmental concerns

Health hazards associated with the use of germicides in health care vary from mucous membrane irritation to death, with the latter involving accidental ingestion by mentally disturbed patients.[126] Although variations exist in the degree of toxicity, as discussed in this document and elsewhere,[4, 127, 128] all disinfectants should be used for the intended purpose only.

Some water and sewer jurisdictions have excluded the disposal of certain chemical germicides (e.g., glutaraldehyde, formaldehyde, phenol) by means of the sewer system. These rules are intended to minimize environmental harm. If hospitals exceed the maximum allowable concentration for a given chemical (e.g., ≤5.0 mg/L), they have three options. First, they can switch to alternative products. For example, they can change from glutaraldehyde to hydrogen peroxide for high-level disinfection or from phenolics to quaternary ammonium compounds for low-level disinfection. Second, the hospitals can collect the disinfectant and dispose of it as a hazardous chemical. Third, they can use a commercially available small-scale treatment system that may neutralize chemicals such as formaldehyde.

European authors have suggested that disinfection by heat rather than chemicals should be used for instruments and ventilation therapy equipment. For example, flushing and washer disinfectors are automated and closed equipment used to 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 with 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 at approximately 90° C, or with steam. Because this machine empties, cleans, and disinfects, manual cleaning is eliminated, fewer disposable items are needed, and less chemical germicides are used. They are available and used in many European countries. Surgical instruments and anesthesia equipment that are more difficult to clean are run in washer-disinfectors with the use of a detergent by use of a longer cycle of 20 to 30 minutes. These machines also disinfect by hot water at approximately 90° C.[129] The stated disadvantages for chemical disinfection include the following: the toxic side effects for the patient caused by chemical residues on the instrument or object; occupational exposure to toxic chemicals; and the danger of recontamination by rinsing the instrument with microbially contaminated tap water.[130]

Transmissible resistance to germicides

Antibiotic resistance among bacteria has been of growing concern in recent years. Of special concern is the increased incidence of infections caused by methicillin-resistant *S. aureus*, vancomycin-resistant *Enterococcus*, multiple-drug-resistant *M. tuberculosis*, and multiple-drug-resistant gram-negative bacilli.

Chromosomal-mediated antibiotic resistance may confer resistance to broad classes of antibiotics (e.g., methicillin-resistant *S. aureus* [MRSA] exhibits resistance to all penicillins and cephalosporins). Many studies have demonstrated that plasmid-mediated resistance may also include multiple drugs. For these reasons, concern has been raised that antibiotic-resistant bacteria might also exhibit cross-resistance to antiseptics and disinfectants.

Several investigators have studied disinfectant resistance in MRSA and methicillin-susceptible *S. aureus* (MSSA). Brumfitt et al.[131] found MRSA more resistant than MSSA strains to chlorhexidine, propamidine, and the quaternary ammonium compound—centrimide. Al-Masaudi et al.[132] reported MRSA and MSSA strains to be equally susceptible to phenols and chlorhexidine but found that MRSA strains were slightly more resistant to quaternary ammonium compounds. Townsend et al.[133-135] demonstrated that a *S. aureus* plasmid carrying gentamicin resistance also encoded resistance to propamidine and quaternary ammonium compounds. Studies have established the involvement of a plasmid locus, *qacA*, in providing protection against quaternary ammonium compounds. Tennant et al.[136, 137] propose that staphylococci evade destruction because the protein specified by the *qacA* determinant is a cytoplasmic membrane–associated protein involved in an efflux system that actively reduces intracellular accumulation in intracellular targets of toxicants such as quaternary ammonium compounds. It has been shown that the presence of the RP1 plasmid in *Escherichia coli* or *P. aeruginosa* does not increase resistance to phenols or quaternary ammonium compounds.[138] Plasmid-mediated resistance to formaldehyde has been demonstrated in *Serratia marcescens*[139] and to hexachlorophene in *P. aeruginosa*.[140]

The literature provides ample evidence of plasmid-mediated resistance to antiseptics and disinfectants. However, these observations have no clinical relevance because even for the more resistant strains the concentrations of disinfectants used in practice are much higher than the observed minimum inhibitory concentrations (MICs). For example, phenolics are used as surface disinfectants at concentrations of approximately 400 ppm and quaternary ammonium compounds at concentrations of approximately 500 ppm. Resistant bacterial strains described in the literature have exhibited MICs less than 15 ppm (μg/ml) for phenolics and quaternary ammonium compounds.[131, 132]

In fact, Rutala et al.[141] found antibiotic-resistant hospital strains of common nosocomial pathogens (i.e., *P. aeruginosa*, *Klebsiella pneumoniae*, *E. coli*, *S. aureus*, *S. epidermidis*, and Enterococcus) to be equally susceptible to disinfectants as antibiotic-sensitive strains by use of the Use-Dilution Method. Other investigators have also been unable to demonstrate a relationship between antibiotic resistance and germicide resistance when the disinfectants are used at the manufacturers' recommended use-dilution. Anderson et al.[142] found similar time-kill curves for vancomycin-resistant and vancomycin-susceptible enterococci by use of a quaternary ammonium compound. Best[143] re-

ported similar inactivation of *M. tuberculosis* and multiple-drug–resistant *M. tuberculosis* (MDR-TB) with 70% ethyl alcohol, 2% glutaraldehyde, 5000 ppm chlorine, and povidone-iodine. Thus vancomycin-resistant enterococcus (VRE), MRSA, and MDR-TB are as sensitive to commonly used hospital disinfectants as drug-sensitive strains at use concentrations.

For these reasons, the CDC does not recommend any special strategies or germicides with higher potencies for cleaning noncritical surfaces in rooms of patients who are infected with multi-antibiotic-resistant organisms such as vancomycin-resistant enterococci. Any EPA-registered germicidal detergent is appropriate for this purpose.[144]

Is there a "double standard" for patient care and processing patient equipment?

Are health care facilities' practices for disinfection consistent in intent and application? For example, semicritical equipment (e.g., endoscopes) should be high-level disinfected between patients; however, some institutions choose to sterilize semicritical equipment when used on certain infectious patients. This may lead to a "double standard" of patient care and is inconsistent with the principle of universal precautions[97] when equipment used on patients with known specific infectious diseases (e.g., tuberculosis, HIV infection) is sterilized, but the same equipment is only high-level–disinfected for other patients. Under these circumstances, sterilization should not be performed in the belief that it is providing a greater margin of safety. In contrast, it is not a double standard of patient care to sterilize endoscopes in one hospital area (e.g., operating room) and high-level disinfect in another area (e.g., gastroenterology clinic) because the outcome is equivalent from an infectious disease transmission perspective.

DISINFECTION

A great number of disinfectants are used in the health care setting, including alcohol, chlorine and chlorine compounds, formaldehyde, glutaraldehyde, hydrogen peroxide, iodophors, phenolics, and quaternary ammonium compounds. These disinfectants are not interchangeable, and the following overview of the performance characteristics of each is intended to provide the user with information to select an appropriate disinfectant and to use it in the safest and most efficient way. It should be recognized that excessive costs may

be attributed to the use of incorrect concentrations and inappropriate germicides. In addition, some disinfectants are formulated in combinations (e.g., hydrogen peroxide and peracetic acid) that may alter their antimicrobial activity. Each formulation of active and inert ingredients is considered a unique product and must undergo the EPA registration approval process, the FDA premarket clearance process, or both. Finally, occupational skin diseases among cleaning personnel have been associated with the use of several disinfectants, such as formaldehyde, glutaraldehyde, chlorine, phenol, and others, and precautions (e.g., gloves, proper ventilation, etc.) should be used to minimize exposure.[145, 146]

Alcohol

In the sphere of hospital disinfection, *alcohol* refers to two water-soluble chemical compounds whose germicidal characteristics are generally underrated; these are *ethyl alcohol* and *isopropyl alcohol*.[147] These alcohols are rapidly bactericidal, rather than bacteriostatic, against vegetative forms of bacteria[148, 149]; they are also tuberculocidal,[147, 150] fungicidal,[34, 151, 152] and virucidal[33, 38, 92-96, 153] but do not destroy bacterial spores. Isopropyl alcohol (20%) has also been shown to be effective in killing the cysts of *Acanthamoeba culbertsoni*.[154] Their cidal activity drops sharply when diluted below 50% concentration, and the optimum bactericidal concentration is in the range of 60% to 90% by volume.[148] The most feasible explanation for the antimicrobial action is denaturation of proteins.

Alcohols are not recommended for sterilizing medical and surgical materials, principally because of their lack of sporicidal action and their inability to penetrate protein-rich materials. Fatal postoperative wound infections with *Clostridium* have occurred when alcohols were used to sterilize surgical instruments contaminated with bacterial spores.[155] Ethyl and isopropyl alcohols are therefore not high-level disinfectants because of their inability to inactivate bacterial spores and because of isopropyl alcohol's inability to kill hydrophilic viruses (e.g., echovirus, coxsackievirus).[38] Alcohols have been used effectively to disinfect oral and rectal thermometers[156, 157] and fiberoptic endoscopes.[158, 159] Alcohol wipes have been used for years to disinfect small surfaces, such as rubber stoppers of multiple-dose medication vials. Furthermore, alcohol is occasionally used to disinfect external surfaces of equipment (e.g., stethoscopes, ventilators, manual ventilation

Table 4. Preparation and stability of chlorine solutions

	Desired chlorine concentration			
	5000 ppm	1000 ppm	500 ppm	100 ppm
Dilution of bleach (5.25% NaOCl) prepared fresh for use within 24 hr	1:10*	1:50	1:100	1:500
Dilution of bleach (5.25% NaOCl) prepared fresh and used for 1-30 days	1:5†	1:25	1:50	1:250

*To achieve a 1:10 dilution, add one part bleach to nine parts water.
†To achieve a 1:5 dilution, add one part bleach to four parts water.

bags[160]), cardiopulmonary resuscitation manikins,[161] or medication preparation areas. Two recent studies demonstrated the effectiveness of 70% isopropyl alcohol to disinfect reusable transducer heads in a controlled environment.[162, 163] In contrast, Beck-Sague and Jarvis[164] described three outbreaks that occurred when alcohol was used to disinfect transducer heads in an intensive care unit setting. The disadvantages of using alcohols on equipment are that they damage the shellac mounting of lensed instruments, tend to cause rubber and certain plastic tubing to swell and harden after prolonged and repeated use, discolor rubber and plastic tiles,[147] and damage tonometer tips (through deterioration of the glue) after the equivalent of 1 working year of routine use.[165] Lingel and Coffey[166] also found that tonometer biprisms soaked in alcohol for 4 days acquired rough front surfaces that could potentially cause corneal damage. This roughening appeared to be caused by a weakening of the cementing substances used to fabricate the biprisms. Corneal opacification has been reported when tonometer tips were swabbed with alcohol immediately before intraocular pressure measurements were taken.[167] Alcohols are flammable and consequently must be stored in a cool, well-ventilated area. They also evaporate rapidly, which makes extended contact times difficult to achieve unless the items are immersed.

Chlorine and chlorine compounds

Hypochlorites, the most widely used of the *chlorine* disinfectants, are available in liquid (e.g., sodium hypochlorite) and solid (e.g., calcium hypochlorite, sodium dichloroisocyanurate) forms. They have a broad spectrum of antimicrobial activity and are inexpensive and fast acting. Use of hypochlorites in hospitals is limited by their corrosiveness, inactivation by organic matter, and relative instability. The microbicidal activity of chlorine is largely attributable to undissociated hy-

pochlorous acid (HOCl). The dissociation of hypochlorous acid to the less microbicidal form (hypochlorite ion, OCl⁻) is dependent on pH. As the pH increases, more hypochlorite ion is formed, and microbicidal activity decreases.[168, 169] A potential hazard is the production of the carcinogen bis-chloromethyl ether when hypochlorite solutions come into contact with formaldehyde[170] and production of the animal carcinogen trihalomethane when hyperchlorinated.[168] A mixture of sodium hypochlorite with acid will also produce a rapid evolution of toxic chlorine gas.

An alternative compound that releases chlorine and is used in the hospital setting is chloramine-T. The advantage of this compound over hypochlorites is that it retains chlorine longer and therefore exerts a more prolonged bactericidal effect. Sodium dichloroisocyanurate tablets are also stable, and the microbicidal activity of solutions prepared from these tablets may be greater than that of sodium hypochlorite solutions containing the same total available chlorine.[171-174]

The exact mechanism by which free chlorine destroys microorganisms has not been elucidated. The postulated mechanism of chlorine disinfection is the inhibition of some key enzymatic reactions within the cell, protein denaturation, and inactivation of nucleic acids.[168]

Low concentrations of free chlorine have biocidal effects on mycoplasma (25 ppm)[175] and vegetative bacteria (< 1 ppm) within seconds in the absence of organic matter.[168] Higher concentrations (1000 ppm) of chlorine are required to kill *M. tuberculosis* according to the AOAC tuberculocidal test.[39] Because household bleach contains 5.25% sodium hypochlorite, or 52,500 ppm available chlorine, a 1:1000 dilution of household bleach provides about 50 ppm available chlorine, and a 1:50 dilution of household bleach provides about 1000 ppm (Table 4). A concentration of 100 ppm will kill 99.9% of *Bacillus subtilis* spores within 5 minutes[176] and

will destroy fungal agents in less than 1 hour.[168] Klein and DeForest[38] reported that 25 different viruses were inactivated in 10 minutes with 200 ppm available chlorine.

Some data are available for chlorine dioxide to substantiate manufacturers' bactericidal, fungicidal, tuberculocidal, sporicidal, and virucidal label claims.[33-35, 39] In 1986, a chlorine dioxide product was voluntarily removed from the market when its use was found to cause dialyzer membrane leaks, which allowed bacteria to migrate from the dialysis fluid side of the dialyzer to the blood side in cellulose-based membranes.[177]

Inorganic chlorine solution is used for disinfecting tonometer heads and for spot disinfection of countertops and floors. A 1:10[10, 178, 179] or 1:100 dilution of 5.25% sodium hypochlorite (household bleach) or an EPA-registered hospital disinfectant[5] can be used for decontamination of blood spills. Either of these methods will minimize the risk of employee exposure to blood. Because hypochlorites and other germicides are substantially inactivated in the presence of blood,[180] the surface should be cleaned before an EPA-registered disinfectant or a 1:10 solution of household bleach is applied (see discussion of OSHA blood-borne pathogen standard). At least 500 ppm available chlorine for 10 minutes is recommended for decontamination of cardiopulmonary resuscitation training manikins.[181] Full-strength bleach has been recommended for the disinfection of needles and syringes in needle-exchange programs for the prevention of blood-borne pathogen spread among intravenous drug–using population. 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.[182] Clinicians should not alter their use of chlorine on surfaces on the basis of testing methods that do not simulate actual disinfection practices.[183]

Chlorine has long been favored as the preferred disinfectant for water treatment. Hyperchlorination of a *Legionella*-contaminated hospital water system resulted in a dramatic decrease (30% to 1.5%) in the isolation of *Legionella pneumophila* from water outlets and a cessation of nosocomial legionnaires' disease in the affected unit.[184] Chloramine T[185] and hypochlorites[186] have been evaluated in disinfecting hydrotherapy equipment.

Hypochlorite solutions in tap water at pH 8.0 or greater are stable for a period of 1 month when stored at room temperature (23° C) in closed, opaque plastic containers.[168, 187] During 1 month at room temperature, the free available chlorine levels of solutions in opened and closed polyethylene containers are reduced maximally to 40% to 50% of the original concentration. On the basis of these data, one investigator recommended that if a user wished to have a solution containing 500 ppm of available chlorine at day 30, a solution initially containing 1000 ppm of chlorine should be prepared (Table 4). After 30 days there was no decomposition of the sodium hypochlorite solution when it was stored in a closed brown bottle.[187]

Formaldehyde

Formaldehyde is used as a disinfectant and a sterilant in both its liquid and its gaseous states. The liquid form will be considered briefly in this section, and a review of formaldehyde as a gas sterilant may be found elsewhere.[188] 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.[38, 189-191] OSHA indicated that formaldehyde should be handled in the workplace as a potential carcinogen, and it set an employee exposure standard for formaldehyde that limits an 8-hour time-weighted average exposure to a concentration of 0.75 ppm.[31, 32] For this reason, employees should have limited direct contact with formaldehyde. These considerations limit the role of formaldehyde in sterilization and disinfection processes.

Formaldehyde inactivates microorganisms by alkylating the amino and sulfhydryl groups of proteins and the ring nitrogen atoms of purine bases.[10] Although formaldehyde-alcohol is a chemical sterilant and formaldehyde is a high-level disinfectant, formaldehyde's hospital uses are limited by its irritating fumes and the pungent odor that is apparent at very low levels (< 1 ppm). For these reasons and others, including carcinogenicity, this germicide is excluded from Table 2. When it is used, direct employee exposure is generally limited; however, significant exposures to formaldehyde have been documented for employees of renal transplant units[192, 193] and students in a gross anatomy laboratory.[194] Formaldehyde is used in the health care setting for preparing viral vaccines (e.g., poliovirus, influenza), as an embalming agent, and for preserving anatomic specimens. In the past it was used, especially as a mixture of formaldehyde and ethanol, for sterilizing

surgical instruments. A survey conducted in 1992 found that formaldehyde was the disinfectant used for reprocessing hemodialyzers by 40% of the hemodialysis centers in the United States, a 54% decrease from 1983.[195] If formaldehyde is used at room temperature, the CDC recommends a concentration of 4% with a minimum exposure time of 24 hours to disinfect disposable hemodialyzers that are reused on the same patient.[196] Aqueous formaldehyde solutions (1% to 2%) have been used to disinfect the internal fluid pathways.[197] To minimize a potential health hazard to patients undergoing dialysis, the dialysis equipment must be thoroughly rinsed and tested for residual formaldehyde before use. Other disinfectants that are available for dialysis systems are chlorine-based disinfectants, glutaraldehyde-based disinfectants, peracetic acid, and peracetic acid with hydrogen peroxide.[196] Some dialysis systems use hot water disinfection for the control of microbial contamination.[196]

Paraformaldehyde

Paraformaldehyde, a solid polymer of formaldehyde, may 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

Glutaraldehyde is a saturated dialdehyde that has gained wide acceptance as a high-level disinfectant and chemical sterilant. Aqueous solutions of glutaraldehyde are acidic and generally in this state are not sporicidal. Only when the solution is "activated" (made alkaline) by alkalizing agents to a pH of 7.5 to 8.5 does the solution become sporicidal. Once activated, these solutions have a shelf life of 14 to 28 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, which are responsible for its biocidal activity.

Novel glutaraldehyde formulations (e.g., glutaraldehyde-phenate, potentiated acid glutaraldehyde, stabilized alkaline glutaraldehyde) have been produced that have overcome the problem of rapid loss of stability (e.g., use life 28 to 30 days) while generally maintaining excellent microbicidal activity.[34, 198-202] It should be realized, however, that antimicrobial activity is dependent not only on age but also on use conditions, such as dilution and organic stress. Manufacturers' litera-

ture for these preparations suggest that the neutral or alkaline glutaraldehydes possess microbicidal and anticorrosion properties superior to those of acid glutaraldehydes. A few published reports substantiate these claims.[203-205] The use of glutaraldehyde-based solutions in hospitals is widespread because of their advantages, which include the following: excellent biocidal properties; activity in the presence of organic matter (20% bovine serum); noncorrosive action on endoscopic equipment, thermometers, and rubber or plastic equipment; and noncoagulation of proteinaceous material.

The biocidal activity of glutaraldehyde is a consequence of its alkylation of sulfhydryl, hydroxyl, carboxyl, and amino groups of microorganisms, which alters RNA, DNA, and protein synthesis.[206]

The in vitro inactivation of microorganisms by glutaraldehydes has been extensively investigated and reviewed.[206] Several investigators showed that 2% aqueous solutions of glutaraldehyde, buffered to a pH of 7.5 to 8.5 with sodium bicarbonate, were effective in killing vegetative bacteria in less than 2 minutes; *M. tuberculosis*, fungi, and viruses in less than 10 minutes; and spores of *Bacillus* and *Clostridium* species in 3 hours.[206-208] Spores of *C. difficile* are more rapidly killed (e.g., 20 minutes) by 2% glutaraldehyde.[46, 110, 111] Concern has been raised about the mycobactericidal prowess of glutaraldehydes because a single investigator using the quantitative suspension test reported that 2% glutaraldehyde inactivated only 2 to 3 logs *M. tuberculosis* in 20 minutes at 20° C.[48] However, all other investigators[15, 39, 41, 42, 49-51] using various test methods, including a quantitative suspension test, have found much greater levels of *M. tuberculosis* inactivation by use of 2% glutaraldehyde. For example, several investigators have demonstrated that glutaraldehyde solutions inactivate 2.4 to >5.0 logs *M. tuberculosis* in 10 minutes (including multidrug-resistant *M. tuberculosis*) and 4.0 to 6.4 logs *M. tuberculosis* at 20 minutes.[15, 39, 41, 42, 49-51] One study reports the isolation of glutaraldehyde-resistant mycobacteria in endoscope washers; however, the clinical significance of this observation is unclear at present.[209] Rubbo et al.[189] showed that 2% alkaline glutaraldehyde has slower action against *M. tuberculosis* than alcohols, formaldehydes, iodine, and phenol. Collins[51] demonstrated that suspensions of *Mycobacterium avium*, *Mycobacterium intracellulare*, and *Mycobacterium gordonae* were more resistant to

disinfection by a 2% alkaline glutaraldehyde (estimated time to sterility 60 minutes) than were virulent *M. tuberculosis* organisms (estimated time to sterility 25 minutes). Collins[51] also showed that the rate of kill was directly proportional to the temperature and the sterility of a standardized suspension of *M. tuberculosis* could not be achieved within 10 minutes. On the basis of these data, 20 minutes at room temperature with a 2% glutaraldehyde is the minimum exposure time needed to reliably kill organisms such as *M. tuberculosis* that are resistant to disinfectants. Glutaraldehyde preparations that are diluted to less than 2% glutaraldehyde should be used as chemical sterilants only after independent verification of their label claims.

There are two publications that evaluate the ability of 2% glutaraldehyde to kill oocytes of *Cryptosporidium* in 30 minutes or 60 minutes.[210, 211] One study found 2% glutaraldehyde to be effective against *Cryptosporidium pavum* at 60 minutes,[210] but another study questioned the ability of glutaraldehyde to kill *Cryptosporidium* in 30 minutes.[211]

Glutaraldehyde is used most commonly as a high-level disinfectant for medical equipment such as endoscopes,[22] respiratory therapy equipment,[212] dialyzers,[213] transducers, anesthesia equipment, spirometry tubing,[214] and hemodialysis proportioning and dialysate delivery systems.[215] Glutaraldehyde is noncorrosive to metal and does not damage lensed instruments, rubber, or plastics. Glutaraldehyde should not be used for cleaning noncritical surfaces; it is too toxic and expensive for this application. Dilution of glutaraldehyde commonly occurs during use. One study showed a glutaraldehyde concentration decline from 2.4% to 1.5% after 10 days in manual and automatic baths used for endoscopes.[216] Others have shown the glutaraldehyde level to fall below 1%, to as low as 0.27%, on day 4 of reuse.[217] These data emphasize the need to ensure that semicritical equipment is disinfected with a minimum effective concentration (MEC) of glutaraldehyde. Most studies suggest that 1.0% glutaraldehyde is the minimum effective concentration when used as a high-level disinfectant,[41, 204, 205] although one investigator using atypical mycobacteria showed that the MEC should be 1.5%.[216] Test strips are available for determining whether an effective concentration of active ingredients (e.g., glutaraldehyde) is present despite repeated use and dilution. The glutaraldehyde test kits have been preliminarily evaluated for accuracy and range,[218, 219] and most test strips are constructed to indicate a concentration above 1.5%. The frequency of testing should be based on how frequently the solutions are used (e.g., if used daily, test daily), but the strip should not be used to extend the use life beyond the expiration date. The solution should be considered unsafe when a dilution of 1% glutaraldehyde or lower is measured.

Proctitis believed to be caused by glutaraldehyde exposure from residual endoscope solution contaminating the air-water channel has been reported and is preventable by thorough endoscope rinsing.[220] Similarly, keratopathy was reported to be caused by ophthalmic instruments that were inadequately rinsed after soaking in 2% glutaraldehyde.[221]

Health care workers can become exposed to elevated levels of glutaraldehyde vapor when equipment is processed in poorly ventilated rooms, when spills occur, or when there are open immersion baths. In these situations, the level of glutaraldehyde in the air could reach its ceiling limit of 0.2 ppm. Engineering and work practice controls that may be used to combat these problems include the following: improved ventilation (7 to 15 air exchanges per hour); use of ducted exhaust hoods or ductless fume hoods with absorbents for glutaraldehyde vapor[222]; tight-fitting lids on immersion baths; and personal protective equipment (e.g., gloves [nitrile rubber, butyl rubber, polyethelyne], goggles) to minimize skin or mucous membrane contact. Some workers have been fitted with a half-face respirator with organic vapor filters[223] or offered a type "C" supplied air respirator with a full facepiece operated in a positive-pressure mode.[224] Even though enforcement of the ceiling limit was suspended on March 23, 1993, by a United States Court of Appeals,[225] it is prudent to limit employee exposure to 0.2 ppm because at this level glutaraldehyde is irritating to the eyes, throat, and nose.[226-228] The American Conference of Governmental Industrial Hygienists issued a "Notice of Intended Changes" in which it was proposed that the ceiling threshold limit value for glutaraldehyde be reduced from 0.2 ppm to 0.05 ppm.[222] Epistaxis, allergic contact dermatitis, asthma, and rhinitis have also been reported in health care workers exposed to glutaraldehyde.[223, 229, 230] Some automated machines for endoscope disinfection reduce employee exposure to glutaraldehyde.[73] Dosimeters are available for measuring glutaraldehyde levels in the workplace.

Hydrogen peroxide

The literature contains limited accounts of the properties, germicidal effectiveness, and potential uses for stabilized *hydrogen peroxide* in the hospital setting. Reports ascribing good germicidal activity to hydrogen peroxide have been published and attest to its bactericidal,[231] virucidal,[232] tuberculocidal,[39] sporicidal,[233] and fungicidal properties.[234] Synergistic sporicidal effects were observed when spores were exposed to a combination of hydrogen peroxide (5.9% to 23.6%) and peracetic acid.[235]

Hydrogen peroxide works by the production of destructive hydroxyl free radicals. These can attack membrane lipids, DNA, and other essential cell components.[234]

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 the disinfection of soft contact lenses (3% for 2 to 3 hours),[234, 236, 237] tonometer biprisms,[166] and ventilators.[238] Corneal damage from a hydrogen peroxide–disinfected tonometer tip that was not properly rinsed has been reported.[239] Hydrogen peroxide has also been instilled into urinary drainage bags in an attempt to eliminate the bag as a source of bladder bacteriuria and environmental contamination.[240, 241] 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.[241]

Concentrations of hydrogen peroxide from 6% to 25% have promise as chemical sterilants. In one recent study, 6% hydrogen peroxide was significantly more effective in the high-level disinfection of the flexible endoscopes than was the 2% glutaraldehyde solution.[73] Hydrogen peroxide has not been widely used for endoscope disinfection, however, because there continues to be concerns that its oxidizing properties may be harmful to some components of the endoscope.[73] The use of hydrogen peroxide for high-level disinfection of semicritical items warrants further study. Chemical irritation resembling pseudomembranous colitis, caused by either 3% hydrogen peroxide or a 2% glutaraldehyde, has been infrequently reported.[242] An epidemic of pseudomembrane-like enteritis and colitis in seven patients in a gastrointestinal endoscopy unit was also associated with use of 3% hydrogen peroxide.[243]

Iodophors

Iodine solutions or tinctures have long been used by health professionals, primarily as antiseptics on skin or tissue. Iodophors, on the other hand, have enjoyed use both as antiseptics and disinfectants. 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, are generally nonstaining and are relatively free of toxicity and irritancy.[244]

Several reports that documented intrinsic microbial contamination of povidone-iodine and poloxamer-iodine[245-247] caused a reappraisal of concepts concerning the chemistry and use of iodophors.[248] It seems that "free" iodine (I_2) is the principal contributor to the bactericidal activity of iodophors, and dilutions of iodophors demonstrate more rapid bactericidal action than a full-strength povidone-iodine solution. The reason that has been suggested for the observation that dilution can increase bactericidal activity is that the dilution of povidone-iodine results in weakening of the iodine linkage to the carrier polymer, with an accompanying increase of free iodine in solution.[246] Iodophor therefore must be used per the manufacturer's recomendations to achieve maximum antimicrobial activity.

Iodine is able to penetrate the cell walls of microorganisms quickly. It is believed that iodine's lethal effects result from a disruption of protein and nucleic acid structure and synthesis.

Published reports on the in vitro antimicrobial efficacy of iodophors demonstrate that iodophors are bactericidal, virucidal, and mycobactericidal but may require prolonged contact times to kill certain fungi and bacterial spores.* Manufacturers' data demonstrate that commercial iodophors are not sporicidal but are tuberculocidal, fungicidal, virucidal, and bactericidal at recommended use dilutions.

In addition to their use as an antiseptic, iodophors have been used for the disinfection of 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 significantly less free iodine than do those formulated as disinfectants.[10]

*References 11, 34, 38, 39, and 249-252.

Peracetic acid

Peracetic acid, or peroxyacetic acid, in low concentrations (0.001% to 0.2%) is characterized by a very rapid action against all microorganisms, including bacterial spores. A special advantage of peracetic acid is that its decomposition products (i.e., acetic acid, water, oxygen, hydrogen peroxide) are not harmful, and it leaves no residue. It remains effective in the presence of organic matter and is sporicidal even at low temperatures. Peracetic acid can corrode copper, brass, bronze, plain steel, and galvanized iron, but these effects can be reduced by additives and pH modification. Peracetic acid 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% to 2% of its activity per month.[253, 254]

Little is known about the mechanism of action of peracetic acid, but it is believed to function in the same manner as other oxidizing agents. It denatures proteins, disrupts the cell wall permeability, and oxidizes sulfhydryl and sulfur bonds in proteins, enzymes, and other metabolites.[253]

The combination of peracetic acid and hydrogen peroxide has been used for disinfecting hemodialyzers.[255] The percentage of centers using a peracetic acid–hydrogen peroxide–based disinfectant for reprocessing dialyzers increased from 5% in 1983 to 52% in 1992.[195] A study showed that patients treated in dialysis units that disinfected dialyzers with a peracetic acid, hydrogen peroxide, acetic acid mixture or with glutaraldehyde had a higher mortality rate than did patients treated in units that used formalin or in units that did not reuse dialyzers. Although the cause of this elevated mortality risk is currently not known, some believe that the germicide is not the causative element, but rather the germicide may be a surrogate indicator of other problems.[256] An automated machine using peracetic acid to chemically process medical, surgical, and dental instruments (e.g., endoscopes, arthroscopes) is used in the United States.[257, 258] Manufacturer's data demonstrated that this system inactivates *Bacillus subtilis* and *Clostridium sporogenes* when the solution is heated to 50° C with an exposure time of 12 minutes or less.[259] Three recent studies have demonstrated that a peracetic acid processor is rapidly sporicidal and bactericidal, and these data suggest the automatic endoscope processor is suitable for processing medical devices such as flexible and rigid scopes.[260-262]

A new product that contains 0.35% peracetic acid has been formulated as a possible alternative to glutaraldehyde and preliminary studies have shown that it has excellent sporicidal and mycobactericical activity.[263, 264]

Phenolics

Phenol (carbolic acid) 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 of the phenol derivatives that are 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 from the parent chemical. Phenolics are assimilated by porous materials, and the residual disinfectant may cause tissue irritation. In 1970 Kahn[265] reported that skin depigmentation is caused by phenolic germicidal detergents containing *para*-tertiary-butylphenol and *para*-tertiary-amylphenol.

At higher 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 the inactivation of essential enzyme systems and leakage of essential metabolites from the cell wall.[266]

Published reports on the antimicrobial efficacy of commonly used phenolic detergents show that phenolics are bactericidal, fungicidal, viricidal, and tuberculocidal.* Data show that three phenolic detergents are bactericidal and tuberculocidal,[268] and another phenol (containing 50% cresol) has little or no virucidal effect against coxsackie B4, echovirus 11, and poliovirus 1.[270] Similarly, Klein and DeForest[38] made the observation that 12% *ortho*-phenylphenol fails to inactivate any of the three hydrophilic viruses after a 10-minute exposure time, although 5% phenol is lethal for these viruses. A 0.5% dilution of a phenolic (2.8% *ortho*-phenylphenol and 2.7% *ortho*-benzyl-*para*-chlorophenol) has been shown to inactivate HIV,[95] and a 2% solution of a phenolic (15% *ortho*-phenylphenol and 6.3% *para*-tertiary-amylphenol) inactivated all but one of 11 fungi tested.[34] Manufacturers' data from tests with the standardized AOAC methods demonstrate that

*References 4, 11, 34, 39, 95, 99, 266-270.

commercial phenolic detergents are not sporicidal but are tuberculocidal, fungicidal, virucidal, and bactericidal at their recommended use dilutions. Generally, these efficacy claims against microorganisms have not been verified by independent laboratories or the EPA. Attempts to substantiate the bactericidal label claims of phenolic detergents with use of the AOAC method have failed.[7,271] These same studies, however, have shown extreme variability of test results among laboratories testing identical products.

This class of compounds is used for decontamination of the hospital environment, including laboratory surfaces, and for noncritical medical and surgical items. Phenolics are not recommended for semicritical items because of the lack of published efficacy data for many of the available formulations and because the residual disinfectant on porous materials may cause tissue irritation even when thoroughly rinsed.

The use of phenolics in nurseries has been justifiably questioned because of the occurrence of hyperbilirubinemia in infants placed in nurseries that use phenolic detergents.[272] In addition, Doan et al.[273] demonstrated microbilirubin level increases in phenolic-exposed infants compared with nonexposed infants when the phenolic was prepared according to the manufacturer's recommended dilution. If phenolics are used to clean nursery floors, they must be diluted according to the recommendation on the product label. Based on these observations, phenolics should not be used to clean infant bassinets and incubators during the stay of an infant. If phenolics are used to terminally clean infant bassinets and incubators, the surfaces should be rinsed thoroughly with water and dried before the infant bassinets and incubators are reused.

Quaternary ammonium compounds

The *quaternary ammonium compounds* have enjoyed wide use as disinfectants and until recently as antiseptics. Benzalkonium chloride ([N]-alkyl [C14 50%, C12 40%, C16 10%] dimethyl benzyl ammonium chloride) was the first commercially available quaternary ammonium compound. This first-generation quaternary ammonium compound, which was introduced in 1935, received acclaim for its microbicidal activity and good detergent action. Common environmental factors, however, such as hard water, soap, anionic residues, and proteinaceous soils, were subsequently found to reduce benzalkonium chloride's effectiveness.

The elimination of such solutions as antiseptics on skin and tissue was recommended by the CDC[1] because of several outbreaks of infections associated with in-use contamination.[274-281] There have also been a few reports of nosocomial infections associated with contaminated quaternary ammonium compounds used to disinfect patient care supplies or equipment such as cystoscopes or cardiac catheters.[279, 282, 283] The quaternary ammonium compounds are good cleaning agents, but materials such as cotton and gauze pads make them less microbicidal because these materials absorb the active ingredients. As with several other germicides (e.g., phenolics, iodophors), gram-negative bacteria have been found to grow in the compounds.[284]

Chemically, the quaternary ammonia compounds are organically substituted ammonium compounds in which the nitrogen atom has a valence of five, four of the substituent radicals (R1 through R4) are alkyl or heterocyclic radicals of a given size or chain length, and the fifth substituent radical (X-) is a halide, sulfate, or similar radical.[285]

Each compound exhibits its own antimicrobial characteristics, so there has been a search for one compound with outstanding antimicrobial properties. The first significant improvement in quaternary ammonium compound technology, referred to as the second-generation quaternary ammonium compound or dual quaternary ammonium compound, was introduced in 1955. The dual quaternary ammonium compound is a combination of ethyl benzyl chloride quaternary ammonium compounds and a modified alkyl chain–distribution dimethyl benzyl ammonium chloride quaternary ammonium compound. Performance in the presence of hard water was purportedly improved.

The third-generation quaternary ammonium compounds, which are referred to as dialkyl or twin-chain quaternary ammonium compounds (such as dodecyl dimethyl ammonium chloride), were introduced in 1965. These quaternary ammonium compounds remained active in hard water and were tolerant of anionic residues.

The bactericidal action of quaternary ammonium compounds has been attributed to inactivation of energy-producing enzymes, denaturation of essential cell proteins, and disruption of the cell membrane. Evidence offered in support of these and other possibilities is provided by Sykes[285] and Petrocci.[286]

Results from manufacturers' data sheets and

from published scientific literature indicate that the quaternary ammonium compounds sold as hospital disinfectants are fungicidal, bactericidal, and virucidal against lipophilic viruses; they are not sporicidal and generally are not tuberculocidal or virucidal against hydrophilic viruses.* Attempts to reproduce the manufacturers' bactericidal and tuberculocidal claims with a limited number of quaternary ammonium compounds by means of the AOAC tests have failed.[7, 39, 271] These same studies, however, showed extreme variability of test results among laboratories testing identical products.

The quaternary ammonium compounds are commonly used in ordinary environmental sanitation of noncritical surfaces such as floors, furniture, and walls.

EMERGING TECHNOLOGIES FOR DISINFECTION AND STERILIZATION

Several other disinfectants and sterilants and sterilization processes are being investigated and may be incorporated into our armamentarium of disinfection and sterilization in the future. The paucity of published studies on disinfectants makes the microbicidal activity of new products difficult to assess. For example, one new high-level disinfectant (*ortho*-phthalaldehyde) requires further evaluation[288] before it can be considered for use on endoscopes.

Reprocessing of heat-labile medical equipment is a major problem in hospitals. ETO has been the sterilant of choice for sterilizing heat-labile medical equipment. Despite ETO's excellent properties, it is toxic, mutagenic, and a suspected carcinogen. Until recently ETO sterilizers combined ETO with a chlorofluorocarbon (CFC) stabilizing agent, most commonly in a ratio of 12% ETO mixed with 88% CFC (referred to as 12/88 ETO). For several reasons health care organizations are exploring the use of new low temperature sterilization technologies.[289] First, CFCs were to be phased out in December 1995 under provisions of the Clean Air Act.[290] 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 by 90% to 99.9%. Third, OSHA regulates the acceptable vapor levels of ETO (i.e., 1 ppm averaged over 8

*References 11, 34, 37, 39, 42, 44, 99, 286, 287.

hours) because of concerns that ETO exposure represents an occupational hazard. These constraints have led to the recent development of alternative technologies for low temperature sterilization in the health care setting.

Alternative technologies to ETO with CFC include the following: 100% ETO; ETO with a different stabilizing gas such as carbon dioxide or hydrochlorofluorocarbons; vaporized hydrogen peroxide; gas plasmas; ozone; and chlorine dioxide. These new technologies should be compared against the characteristics of an ideal low-temperature (<60° C) sterilant.[289] Although it is apparent that all technologies will have limitations,[291] understanding the limitations imposed by restrictive device designs (e.g., long, narrow lumens) is critical for proper application of new sterilization technology.[292] 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. There are peer-reviewed scientific data demonstrating concerns about the efficacy of several of the low-temperature sterilization processes (i.e., gas plasma, vaporized hydrogen peroxide, ETO), particularly when the test organisms are challenged in the presence of serum and salt and a narrow lumen vehicle.[292, 294]

RECOMMENDATIONS

A. Cleaning, disinfecting, and sterilizing patient care equipment: All objects to be high-level disinfected or sterilized should first be thoroughly cleaned to remove all organic matter (e.g., blood, tissue) and other residue.

B. Indications for sterilization and high-level disinfection (recommendations B.1. and B.4. per 1985 CDC guideline[2] and recommendation B.5 per 1993 CDC guideline).[88]

1. Critical medical devices or pieces of patient care equipment that enter normally sterile tissue or the vascular system or through which blood flows should be sterilized before each use.

2. Endoscope accessories: Biopsy forceps or other cutting instruments that break the mucosal barrier should be sterilized. Other endoscope accessories (e.g., suction valves) should be sterilized after each patient use; if this is not feasible, they should receive at least high-level disinfection. Please refer to the "APIC Guideline for Infection Preven-

tion and Control in Flexible Endoscopy" for additional recommendations.[23]

3. Laparoscopes, arthroscopes, and other scopes that enter normally sterile tissue should be subjected to a sterilization procedure before each use; if this is not feasible, they should receive at least high-level disinfection. Disinfection should be followed by a rinse with sterile water.

4. Equipment that touches mucous membranes (e.g., endoscopes, endotracheal tubes, anesthesia breathing circuits, and respiratory therapy equipment) should receive high-level disinfection.

5. Dental instruments that penetrate soft tissue or bone (e.g., forceps, scalpels, bone chisels, scalers, and burs) are classified as critical and should be sterilized or discarded after each use. Dental instruments that are not intended to penetrate oral soft tissue or bone (e.g., amalgam condensers, air-water syringes) but may come into contact with oral tissues are classified as semicritical and should be sterilized after each use. If the semicritical instrument could be damaged by the sterilization process, the instrument should be high-level disinfected. Noncritical surfaces, such as uncovered operatory surfaces (e.g., countertops, chair switches), should be disinfected between patients with an intermediate-level or low-level disinfectant.

C. Chemical methods for sterilization (Table 2): When sterilization is indicated and other sterilization methods (e.g., steam or ETO) cannot be used, any one of three liquid chemical sterilants (see Table 2) may be used. The manufacturer's instructions for use will specify the recommended exposure time.

D. Selection and use of high-level disinfectants for semicritical patient care items.

1. Solutions containing glutaraldehyde, hydrogen peroxide, chlorine, and peracetic acid can achieve high-level disinfection if objects are properly cleaned before disinfection. See Table 2 for recommended concentrations. The disinfectant or chemical sterilant selected should have no or minimal deleterious effects on the object (e.g., chlorine may corrode metals; see Table 2).

2. The exact time for disinfecting semicritical items is somewhat elusive at present because of conflicting label claims and lack of agreement in published literature, espe-

cially regarding the mycobactericidal activity of glutaraldehydes. The longer the exposure of an item to a disinfectant, the more likely it is that all contaminating microorganisms will be inactivated. Unfortunately, with extended exposure to a disinfectant it is also more likely that delicate and intricate instruments such as endoscopes may be damaged. Medical equipment such as endoscopes, which are difficult to clean and disinfect because of narrow channels or other areas that can harbor organisms (e.g., crevices, joints), should be exposed to a high-level disinfectant for at least 20 minutes at room temperature after cleaning.

E. Selection and use of low-level disinfectants for noncritical patient care items.

1. Solutions for use on noncritical patient care equipment and recommended concentrations are listed in Table 2.

2. The contact time is 10 minutes or less.

3. Phenolics should not be used to clean infant bassinets and incubators during the stay of an infant. If phenolics are used to terminally clean infant bassinets and incubators, the surfaces should be rinsed thoroughly with water and dried before the infant bassinets and incubators are reused.

F. Processing patient care equipment contaminated with HIV or HBV.

1. Standard sterilization and disinfection procedures for patient care equipment (as recommended in this guideline) are adequate to sterilize or disinfect instruments or devices contaminated with blood or other body fluids from persons infected with blood-borne pathogens, including HIV. No changes in procedures for cleaning, disinfecting, or sterilizing need to be made.

2. Noncritical environmental surfaces contaminated with blood or bloody body fluids should be cleaned before an EPA-registered disinfectant/detergent is applied for disinfection. Persons cleaning spills should wear disposable gloves and other personal protective equipment as indicated.

G. Processing CJD-contaminated patient care equipment

1. The only infectious agent that requires unique decontamination recommendations is the CJD prion. The need for such recommendations is due to an extremely resistant subpopulation of prions and the protection afforded this tissue-associated agent.

2. Critical and semicritical CJD-contaminated care equipment should preferably be steam sterilized for at least 30 minutes at a temperature of 132° C (121° C is not effective) in a gravity displacement sterilizer. A prevacuum sterilizer used for 18 minutes at 134° C to 138° C has also been found to be effective. Immersion in 1 N sodium hydroxide (which is caustic) for 1 hour at room temperature followed by steam sterilization at 121° C for 30 minutes is an alternative procedure for critical and semicritical items. Because noncritical patient care items or surfaces (e.g., autopsy tables, floors) have not been involved in disease transmission, these surfaces may be disinfected with either bleach (undiluted, or up to 1:10 dilution) or 1 N sodium hydroxide at room temperature for 15 minutes or less. A formalin–formic acid procedure is required for inactivating virus infectivity in tissue samples from patients with CJD.

H. Method of processing reusable transducers: After transducers are cleaned, they may be sterilized with ETO or disinfected with a high-level disinfectant. Alternatively, transducer heads may be disinfected with 70% isopropyl alcohol. However, the disinfection procedure must be adhered to rigorously, and this is best accomplished in a controlled setting. The transducers should be stored in a manner to prevent recontamination before use.

I. The selection and use of disinfectants in the health care field is dynamic, and products may become available that were not in existence when this guideline was written. As newer disinfectants become available, persons or committees responsible for selecting disinfectants should be guided by information in the scientific literature.

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