Guidelines for Environmental Infection Control in Health-Care Facilities

Part 2

D. Water

1. Modes of Transmission of Waterborne Diseases

Moist environments and aqueous solutions in health-care settings have the potential to serve as reservoirs for waterborne microorganisms. Under favorable environmental circumstances (e.g., warm temperature and the presence of a source of nutrition), many bacterial and some protozoal microorganisms can either proliferate in active growth or remain for long periods in highly stable, environmentally resistant (yet infectious) forms. Modes of transmission for waterborne infections

include a) direct contact [e.g., that required for hydrotherapy]; b) ingestion of water [e.g., through consuming contaminated ice]; c) indirect-contact transmission [e.g., from an improperly reprocessed medical device];[6] d) inhalation of aerosols dispersed from water sources;[3] and e) aspiration of contaminated water. The first three modes of transmission are commonly associated with infections caused by gram-negative bacteria and nontuberculous mycobacteria (NTM). Aerosols generated from water sources contaminated with *Legionella* spp. often serve as the vehicle for introducing legionellae to the respiratory tract.[394]

2. Waterborne Infectious Diseases in Health-Care Facilities

a. Legionellosis

Legionellosis is a collective term describing infection produced by *Legionella* spp., whereas Legionnaires disease is a multi-system illness with pneumonia.[395] The clinical and epidemiologic aspects of these diseases (Table 11) are discussed extensively in another guideline.[3] Although Legionnaires disease is a respiratory infection, infection-control measures intended to prevent health-care–associated cases center on the quality of water—the principal reservoir for *Legionella* spp.

Table 11. Clinical and epidemiologic characteristics of legionellosis/Legionnaires disease

		References
Causative agent	***Legionella pneumophila*** (90% of infections); *L. micdadei, L. bozemanii, L. dumoffii, L. longbeachii,* (14 additional species can cause infection in humans)	395–399
Mode of transmission	Aspiration of water, direct inhalation or water aerosols	3, 394–398, 400
Source of exposure	Exposure to environmental sources of *Legionella* spp. (i.e., water or water aerosols)	31, 33, 401–414
Clinical syndromes and diseases	Two distinct illnesses: a) Pontiac fever [a milder, influenza-like illness]; and b) progressive **pneumonia** that may be accompanied by cardiac, renal, and gastrointestinal involvement	3, 397–399, 415–422
Populations at greatest risk	Immunosuppressed patients (e.g., transplant patients, cancer patients, and patients receiving corticosteroid therapy); immunocompromised patients (e.g., surgical patients, patients with underlying chronic lung disease, and dialysis patients); elderly persons; and patients who smoke	395–397, 423–433
Occurrence	Proportion of community-acquired pneumonia caused by *Legionella* spp. ranges from 1%–5%; estimated annual incidence among the general population is 8,000–18,000 cases in the United States; the incidence of health-care–associated pneumonia (0%–14%) may be underestimated if appropriate laboratory diagnostic methods are unavailable.	396, 397, 434–444
Mortality rate	Mortality declined markedly during 1980–1998, from 34% to 12% for all cases; the mortality rate is higher among persons with health-care–associated pneumonia compared with the rate among community-acquired pneumonia patients (14% for health-care–associated pneumonia versus 10% for community-acquired pneumonia [1998 data]).	395–397, 445

Legionella spp. are commonly found in various natural and man-made aquatic environments[446, 447] and can enter health-care facility water systems in low or undetectable numbers.[448, 449] Cooling towers, evaporative condensers, heated potable water distribution systems, and locally-produced distilled water can provide environments for multiplication of legionellae.[450–454] In several hospital outbreaks, patients have been infected through exposure to contaminated aerosols generated by cooling towers, showers, faucets, respiratory therapy equipment, and room-air humidifiers.[401–410, 455] Factors that enhance

colonization and amplification of legionellae in man-made water environments include a) temperatures of 77°F–107.6°F [25°C–42°C],[456–460] b) stagnation,[461] c) scale and sediment, [462] and d) presence of certain free-living aquatic amoebae that can support intracellular growth of legionellae.[462, 463] The bacteria multiply within single-cell protozoa in the environment and within alveolar macrophages in humans.

b. Other Gram-Negative Bacterial Infections

Other gram-negative bacteria present in potable water also can cause health-care–associated infections. Clinically important, opportunistic organisms in tap water include *Pseudomonas aeruginosa, Pseudomonas* spp., *Burkholderia cepacia, Ralstonia pickettii, Stenotrophomonas maltophilia*, and *Sphingomonas* spp. (Tables 12 and 13). Immunocompromised patients are at greatest risk of developing infection. Medical conditions associated with these bacterial agents range from colonization of the respiratory and urinary tracts to deep, disseminated infections that can result in pneumonia and bloodstream bacteremia. Colonization by any of these organisms often precedes the development of infection. The use of tap water in medical care (e.g., in direct patient care, as a diluent for solutions, as a water source for medical instruments and equipment, and during the final stages of instrument disinfection) therefore presents a potential risk for exposure. Colonized patients also can serve as a source of contamination, particularly for moist environments of medical equipment (e.g., ventilators).

In addition to *Legionella* spp., *Pseudomonas aeruginosa* and *Pseudomonas* spp. are among the most clinically relevant, gram-negative, health-care–associated pathogens identified from water. These and other gram-negative, non-fermentative bacteria have minimal nutritional requirements (i.e., these organisms can grow in distilled water) and can tolerate a variety of physical conditions. These attributes are critical to the success of these organisms as health-care–associated pathogens. Measures to prevent the spread of these organisms and other waterborne, gram-negative bacteria include hand hygiene, glove use, barrier precautions, and eliminating potentially contaminated environmental reservoirs.[464, 465]

Table 12. *Pseudomonas aeruginosa* infections in health-care facilities

		References
Clinical syndromes and diseases	Septicemia, pneumonia (particularly ventilator-associated), chronic respiratory infections among cystic fibrosis patients, urinary tract infections, skin and soft-tissue infections (e.g., tissue necrosis and hemorrhage), burn-wound infections, folliculitis, endocarditis, central nervous system infections (e.g., meningitis and abscess), eye infections, and bone and joint infections	466–503
Modes of transmission	Direct contact with water, aerosols; aspiration of water and inhalation of water aerosols; and indirect transfer from moist environmental surfaces via hands of health-care workers	28, 502–506
Environmental sources of pseudomonads in health-care settings	Potable (tap) water, distilled water, antiseptic solutions contaminated with tap water, sinks, hydrotherapy pools, whirlpools and whirlpool spas, water baths, lithotripsy therapy tanks, dialysis water, eyewash stations, flower vases, and endoscopes with residual moisture in the channels	28, 29, 466, 468, 507–520
Environmental sources of pseudomonads in the community	Fomites (e.g., drug injection equipment stored in contaminated water)	494, 495
Populations at greatest risk	Intensive care unit (ICU) patients (including neonatal ICU), transplant patients (organ and hematopoietic stem cell), neutropenic patients, burn therapy and hydrotherapy patients, patients with malignancies, cystic fibrosis patients, patients with underlying medical conditions, and dialysis patients	28, 466, 467, 472, 477, 493, 506–508, 511, 512, 521–526

Table 13. Other gram-negative bacteria associated with water and moist environments

Implicated contaminated environmental vehicle	References
Burkholderia cepacia	
Distilled water	527
Contaminated solutions and disinfectants	528, 529
Dialysis machines	527
Nebulizers	530–532
Water baths	533
Intrinsically-contaminated mouthwash*	534
Ventilator temperature probes	535
***Stenotrophomonas maltophlia, Sphingomonas* spp.**	
Distilled water	536, 537
Contaminated solutions and disinfectants	529
Dialysis machines	527
Nebulizers	530–532
Water	538
Ventilator temperature probes	539
Ralstonia pickettii	
Fentanyl solutions	540
Chlorhexidine	541
Distilled water	541
Contaminated respiratory therapy solution	541, 542
Serratia marcescens	
Potable water	543
Contaminated antiseptics (i.e., benzalkonium chloride and chlorhexidine)	544–546
Contaminated disinfectants (i.e., quaternary ammonium compounds and glutaraldehyde)	547, 548
***Acinetobacter* spp.**	
Medical equipment that collects moisture (e.g., mechanical ventilators, cool mist humidifiers, vaporizers, and mist tents)	549–556
Room humidifiers	553, 555
Environmental surfaces	557–564
***Enterobacter* spp.**	
Humidifier water	565
Intravenous fluids	566–578
Unsterilized cotton swabs	573
Ventilators	565, 569
Rubber piping on a suctioning machine	565, 569
Blood gas analyzers	570

* This report describes intrinsic contamination (i.e., occurring during manufacture) prior to use by the health-care facility staff. All other entries reflect extrinsic sources of contamination.

Two additional gram-negative bacterial pathogens that can proliferate in moist environments are *Acinetobacter* spp. and *Enterobacter* spp.[571, 572] Members of both genera are responsible for health-care–associated episodes of colonization, bloodstream infections, pneumonia, and urinary tract infections among medically compromised patients, especially those in ICUs and burn therapy units.[566, 572–583] Infections caused by *Acinetobacter* spp. represent a significant clinical problem. Average infection rates are higher from July through October compared with rates from November through June.[584] Mortality rates associated with *Acinetobacter* bacteremia are 17%–52%, and rates as high as 71% have been reported for pneumonia caused by infection with either *Acinetobacter* spp. or

Pseudomonas spp.[574–576] Multi-drug resistance, especially in third generation cephalosporins for *Enterobacter* spp., contributes to increased morbidity and mortality.[569, 572]

Patients and health-care workers contribute significantly to the environmental contamination of surfaces and equipment with *Acinetobacter* spp. and *Enterobacter* spp., especially in intensive care areas, because of the nature of the medical equipment (e.g., ventilators) and the moisture associated with this equipment.[549, 571, 572, 585] Hand carriage and hand transfer are commonly associated with health-care–associated transmission of these organisms and for *S. marcescens*.[586] *Enterobacter* spp. are primarily spread in this manner among patients by the hands of health-care workers.[567, 587] *Acinetobacter* spp. have been isolated from the hands of 4%–33% of health-care workers in some studies,[585–590] and transfer of an epidemic strain of *Acinetobacter* from patients' skin to health-care workers' hands has been demonstrated experimentally.[591] *Acinetobacter* infections and outbreaks have also been attributed to medical equipment and materials (e.g., ventilators, cool mist humidifiers, vaporizers, and mist tents) that may have contact with water of uncertain quality (e.g., rinsing a ventilator circuit in tap water).[549–556] Strict adherence to hand hygiene helps prevent the spread of both *Acinetobacter* spp. and *Enterobacter* spp.[577, 592]

Acinetobacter spp. have also been detected on dry environmental surfaces (e.g., bed rails, counters, sinks, bed cupboards, bedding, floors, telephones, and medical charts) in the vicinity of colonized or infected patients; such contamination is especially problematic for surfaces that are frequently touched.[557–564] In two studies, the survival periods of *Acinetobacter baumannii* and *Acinetobacter calcoaceticus* on dry surfaces approximated that for *S. aureus* (e.g., 26–27 days).[593, 594] Because *Acinetobacter* spp. may come from numerous sources at any given time, laboratory investigation of health-care–associated *Acinetobacter* infections should involve techniques to determine biotype, antibiotype, plasmid profile, and genomic fingerprinting (i.e., macrorestriction analysis) to accurately identify sources and modes of transmission of the organism(s).[595]

c. Infections and Pseudo-Infections Due to Nontuberculous Mycobacteria

NTM are acid-fast bacilli (AFB) commonly found in potable water. NTM include both saprophytic and opportunistic organisms. Many NTM are of low pathogenicity, and some measure of host impairment is necessary to enhance clinical disease.[596] The four most common forms of human disease associated with NTM are a) pulmonary disease in adults; b) cervical lymph node disease in children; c) skin, soft tissue, and bone infections; and d) disseminated disease in immunocompromised patients.[596, 597] Person-to-person acquisition of NTM infection, especially among immunocompetent persons, does not appear to occur, and close contacts of patients are not readily infected, despite the high numbers of organisms harbored by such patients.[596, 598–600] NTM are spread via all modes of transmission associated with water. In addition to health-care–associated outbreaks of clinical disease, NTM can colonize patients in health-care facilities through consumption of contaminated water or ice or through inhalation of aerosols.[601–605] Colonization following NTM exposure, particularly of the respiratory tract, occurs when a patient's local defense mechanisms are impaired; overt clinical disease does not develop.[606] Patients may have positive sputum cultures in the absence of clinical disease.

Using tap water during patient procedures and specimen collection and in the final steps of instrument reprocessing can result in pseudo-outbreaks of NTM contamination.[607–609] NTM pseudo-outbreaks of *Mycobacterium chelonae, M. gordonae,* and *M. xenopi* have been associated with both bronchoscopy and gastrointestinal endoscopy when a) tap water is used to provide irrigation to the site or to rinse off the viewing tip *in situ* or b) the instruments are inappropriately reprocessed with tap water in the final steps.[610–612]

Table 14. Nontuberculous mycobacteria—environmental vehicles

Vehicles associated with infections or colonizations	References
Mycobacterium abscessus	
Inadequately sterilized medical instruments	613
***Mycobacterium avium* complex (MAC)**	
Potable water	614–616
Mycobacterium chelonae	
Dialysis, reprocessed dialyzers	31, 32
Inadequately-sterilized medical instruments, jet injectors	617, 618
Contaminated solutions	619, 620
Hydrotherapy tanks	621
Mycobacterium fortuitum	
Aerosols from showers or other water sources	605, 606
Ice	602
Inadequately sterilized medical instruments	603
Hydrotherapy tanks	622
Mycobacterium marinum	
Hydrotherapy tanks	623
Mycobacterium ulcerans	
Potable water	624

Vehicles associated with pseudo-outbreaks	References
Mycobacterium chelonae	
Potable water used during bronchoscopy and instrument reprocessing	610
Mycobacterium fortuitum	
Ice	607
Mycobacterium gordonae	
Deionized water	611
Ice	603
Laboratory solution (intrinsically contaminated)	625
Potable water ingestion prior to sputum specimen collection	626
Mycobacterium kansasii	
Potable water	627
Mycobacterium terrae	
Potable water	608
Mycobacterium xenopi	
Potable water	609, 612, 627

NTM can be isolated from both natural and man-made environments. Numerous studies have identified various NTM in municipal water systems and in hospital water systems and storage tanks.[615, 616, 624, 627–632] Some NTM species (e.g., *Mycobacterium xenopi*) can survive in water at 113°F (45°C), and can be isolated from hot water taps, which can pose a problem for hospitals that lower the temperature of their hot water systems.[627] Other NTM (e.g., *Mycobacterium kansasii, M. gordonae, M. fortuitum*, and *M. chelonae*) cannot tolerate high temperatures and are associated more often with cold water lines and taps.[629]

NTM have a high resistance to chlorine; they can tolerate free chlorine concentrations of 0.05–0.2 mg/L (0.05–0.2 ppm) found at the tap.[598, 633, 634] They are 20–100 times more resistant to chlorine compared with coliforms; slow-growing strains of NTM (e.g., *Mycobacterium avium* and *M. kanasii*) appear to be

more resistant to chorine inactivation compared to fast-growing NTM.[635] Slow-growing NTM species have also demonstrated some resistance to formaldehyde and glutaraldehyde, which has posed problems for reuse of hemodialyzers.[31] The ability of NTM to form biofilms at fluid-surface interfaces (e.g., interior surfaces of water pipes) contributes to the organisms' resistance to chemical inactivation and provides a microenvironment for growth and proliferation.[636, 637]

d. Cryptosporidiosis

Cryptosporidium parvum is a protozoan parasite that causes self-limiting gastroenteritis in normal hosts but can cause severe, life-threatening disease in immunocompromised patients. First recognized as a human pathogen in 1976, *C. parvum* can be present in natural and finished waters after fecal contamination from either human or animal sources.[638–641]

The health risks associated with drinking potable water contaminated with minimal numbers of *C. parvum* oocysts are unknown.[642] It remains to be determined if immunosuppressed persons are more susceptible to lower doses of oocysts than are immunocompetent persons. One study demonstrated that a median 50% infectious dose (ID_{50}) of 132 oocysts of calf origin was sufficient to cause infection among healthy volunteers.[643] In a second study, the same researchers found that oocysts obtained from infected foals (newborn horses) were infectious for human volunteers at median ID_{50} of 10 oocysts, indicating that different strains or species of *Cryptosporidium* may vary in their infectivity for humans.[644] In a small study population of 17 healthy adults with pre-existing antibody to *C. parvum*, the ID_{50} was determined to be 1,880 oocysts, more than 20-fold higher than in seronegative persons.[645] These data suggest that pre-existing immunity derived from previous exposures to *Cryptosporidium* offers some protection from infection and illness that ordinarily would result from exposure to low numbers of oocysts.[645, 646]

Oocysts, particularly those with thick walls, are environmentally resistant, but their survival under natural water conditions is poorly understood. Under laboratory conditions, some oocysts remain viable and infectious in cold (41°F [5°C]) for months.[641] The prevalence of *Cryptosporidium* in the U.S. drinking water supply is notable. Two surveys of approximately 300 surface water supplies revealed that 55%–77% of the water samples contained *Cryptosporidium* oocysts.[647, 648] Because the oocysts are highly resistant to common disinfectants (e.g., chlorine) used to treat drinking water, filtration of the water is important in reducing the risk of waterborne transmission. Coagulation-floculation and sedimentation, when used with filtration, can collectively achieve approximately a 2.5 $\log_{10}$ reduction in the number of oocysts.[649] However, outbreaks have been associated with both filtered and unfiltered drinking water systems (e.g., the 1993 outbreak in Milwaukee, Wisconsin that affected 400,000 people).[641, 650–652] The presence of oocysts in the water is not an absolute indicator that infection will occur when the water is consumed, nor does the absence of detectable oocysts guarantee that infection will not occur. Health-care–associated outbreaks of cryptosporidiosis primarily have been described among groups of elderly patients and immunocompromised persons.[653]

3. Water Systems in Health-Care Facilities

a. Basic Components and Point-of-Use Fixtures

Treated municipal water enters a health-care facility via the water mains and is distributed throughout the building(s) by a network of pipes constructed of galvanized iron, copper, and polyvinylchloride (PVC). The pipe runs should be as short as is practical. Where recirculation is employed, the pipe runs should be insulated and long dead legs avoided in efforts to minimize the potential for water stagnation, which favors the proliferation of *Legionella* spp. and NTM. In high-risk applications (e.g., PE areas for severely immunosuppressed patients), insulated recirculation loops should be incorporated as a design

feature. Recirculation loops prevent stagnation and insulation maintains return water temperature with minimal loss.

Each water service main, branch main, riser, and branch (to a group of fixtures) has a valve and a means to reach the valves via an access panel.[120] Each fixture has a stop valve. Valves permit the isolation of a portion of the water system within a facility during repairs or maintenance. Vacuum breakers and other similar devices in the lines prevent water from back-flowing into the system. All systems that supply water should be evaluated to determine risk for potential back siphonage and cross connections.

Health-care facilities generate hot water from municipal water using a boiler system. Hot water heaters and storage vessels for such systems should have a drainage facility at the lowest point, and the heating element should be located as close as possible to the bottom of the vessel to facilitate mixing and to prevent water temperature stratification. Those hot or cold water systems that incorporate an elevated holding tank should be inspected and cleaned annually. Lids should fit securely to exclude foreign materials.

The most common point-of-use fixtures for water in patient-care areas are sinks, faucets, aerators, showers, and toilets; eye-wash stations are found primarily in laboratories. The potential for these fixtures to serve as a reservoir for pathogenic microorganisms has long been recognized (Table 15).[509, 654–656] Wet surfaces and the production of aerosols facilitate the multiplication and dispersion of microbes. The level of risk associated with aerosol production from point-of-use fixtures varies. Aerosols from shower heads and aerators have been linked to a limited number of clusters of gram-negative bacterial colonizations and infections, including Legionnaires disease, especially in areas where immunocompromised patients are present (e.g., surgical ICUs, transplant units, and oncology units).[412, 415, 656–659] In one report, clinical infection was not evident among immunocompetent persons (e.g., hospital staff) who used hospital showers when *Legionella pneumophila* was present in the water system.[660] Given the infrequency of reported outbreaks associated with faucet aerators, consensus has not been reached regarding the disinfection of or removal of these devices from general use. If additional clusters of infections or colonizations occur in high-risk patient-care areas, it may be prudent to clean and decontaminate the aerators or to remove them.[658, 659] ASHRAE recommends cleaning and monthly disinfection of aerators in high-risk patient-care areas as part of *Legionella* control measures.[661] Although aerosols are produced with toilet flushing,[662, 663] no epidemiologic evidence suggests that these aerosols pose a direct infection hazard.

Although not considered a standard point-of-use fixture, decorative fountains are being installed in increasing numbers in health-care facilities and other public buildings. Aerosols from a decorative fountain have been associated with transmission of *Legionella pneumophila* serogroup 1 infection to a small cluster of older adults.[664] This hotel lobby fountain had been irregularly maintained, and water in the fountain may have been heated by submersed lighting, all of which favored the proliferation of *Legionella* in the system.[664] Because of the potential for generations of infectious aerosols, a prudent prevention measure is to avoid locating these fixtures in or near high-risk patient-care areas and to adhere to written policies for routine fountain maintenance.[120]

Table 15. Water and point-of-use fixtures as sources and reservoirs of waterborne pathogens*

Reservoir	Associated pathogens	Transmission	Strength of evidence+	Prevention and control	References
Potable water	*Pseudomonas*, gram-negative bacteria, NTM	Contact	Moderate	Follow public health guidelines.	(See Tables 12–14)

Reservoir	Associated pathogens	Transmission	Strength of evidence+	Prevention and control	References
Potable water	*Legionella*	Aerosol inhalation	Moderate	Provide supplemental treatment for water.	(See Table 11)
Holy water	Gram-negative bacteria	Contact	Low	Avoid contact with severe burn injuries. Minimize use among immunocompromised patients.	665
Dialysis water	Gram-negative bacteria	Contact	Moderate	Dialysate should be ≤2,000 cfu/mL; water should be ≤200 cfu/mL.	2, 527, 666–668
Automated endoscope reprocessors and rinse water	Gram-negative bacteria	Contact	Moderate	Use and maintain equipment according to instructions; eliminate residual moisture by drying the channels (e.g., through alcohol rinse and forced air drying).	669–675
Water baths	*Pseudomonas, Burkholderia, Acinetobacter*	Contact	Moderate	Add germicide to the water; wrap transfusion products in protective plastic wrap if using the bath to modulate the temperature of these products.	29, 533, 676, 677
Tub immersion	*Pseudomonas, Enterobacter, Acinetobacter*	Contact	Moderate	Drain and disinfect tub after each use; consider adding germicide to the water; water in large hydrotherapy pools should be properly disinfected and filtered.	678–683
Ice and ice machines	NTM, *Enterobacter, Pseudomonas, Cryptosporidium* *Legionella*	Ingestion, contact	Moderate Low	Clean periodically; use automatic dispenser (avoid open chest storage compartments in patient areas).	601, 684–687
Faucet aerators	*Legionella*	Aerosol inhalation	Moderate	Clean and disinfect monthly in high-risk patient areas; consider removing if additional infections occur.	415, 661
Faucet aerators	*Pseudomonas, Acinetobacter, Stenotrophomonas, Chryseobacterium*	Contact, droplet	Low	No precautions are necessary at present in immunocompetent patient-care areas.	658, 659, 688, 689
Sinks	*Pseudomonas*	Contact, droplet	Moderate	Use separate sinks for handwashing and disposal of contaminated fluids.	509, 653, 685–693
Showers	*Legionella*	Aerosol inhalation	Low	Provide sponge baths for hematopoietic stem cell transplant patients; avoid shower use for immunocompromised patients when *Legionella* is detected in facility water.	656

Reservoir	Associated pathogens	Transmission	Strength of evidence+	Prevention and control	References
Dental unit water lines	*Pseudomonas, Legionella, Sphingomonas, Acinetobacter*	Contact	Low	Clean water systems according to system manufacturer's instructions.	636, 694–696
Ice baths for thermodilution catheters	*Ewingella, Staphylococcus*	Contact	Low	Use sterile water.	697, 698
Decorative fountains	*Legionella*	Aerosol inhalation	Low	Perform regular maintenance, including water disinfection; avoid use in or near high-risk patient-care areas.	664
Eyewash stations	*Pseudomonas,* amoebae, *Legionella*	Contact	Low Minimum	Flush eyewash stations weekly; have sterile water available for eye flushes.	518, 699, 700
Toilets	Gram-negative bacteria	–	Minimum	Clean regularly; use good hand hygiene.	662
Flowers	Gram-negative bacteria, *Aspergillus*	–	Minimum	Avoid use in intensive care units and in immunocompromised patient-care settings.	515, 701, 702

* Modified from reference 654 and used with permission of the publisher (Slack, Inc.)
\+ **Moderate:** occasional well-described outbreaks. **Low:** few well-described outbreaks. **Minimal:** actual infections not demonstrated.

b. Water Temperature and Pressure

Hot water temperature is usually measured at the point of use or at the point at which the water line enters equipment requiring hot water for proper operation.[120] Generally, the hot water temperature in hospital patient-care areas is no greater than a temperature within the range of 105°F–120°F (40.6°C–49°C), depending on the AIA guidance issued at the year in which the facility was built.[120] Hot water temperature in patient-care areas of skilled nursing-care facilities is set within a slightly lower range of 95°F–110°F (35°C–43.3°C) depending on the AIA guidance at the time of facility construction.[120] Many states have adopted a temperature setting in these ranges into their health-care regulations and building codes. ASHRAE, however, has recommended higher settings.[661] Steam jets or booster heaters are usually needed to meet the hot water temperature requirements in certain service areas of the hospital (e.g., the kitchen [120°F (49°C)] or the laundry [160°F (71°C)]).[120] Additionally, water lines may need to be heated to a particular temperature specified by manufacturers of specific hospital equipment. Hot-water distribution systems serving patient-care areas are generally operated under constant recirculation to provide continuous hot water at each hot-water outlet.[120] If a facility is or has a hemodialysis unit, then continuously circulated, cold treated water is provided to that unit.[120]

To minimize the growth and persistence of gram-negative waterborne bacteria (e.g., thermophilic NTM and *Legionella* spp.),[627, 703–709] cold water in health-care facilities should be stored and distributed at temperatures below 68°F (20°C); hot water should be stored above 140°F (60°C) and circulated with a minimum return temperature of 124°F (51°C),[661] or the highest temperature specified in state regulations and building codes. If the return temperature setting of 124°F (51°C) is permitted, then installation of preset thermostatic mixing valves near the point-of-use can help to prevent scalding. Valve maintenance is especially important in preventing valve failure, which can result in scalding. New shower systems in large buildings, hospitals, and nursing homes should be designed to permit mixing of hot and cold water near the shower head. The warm water section of pipe between the control valve and shower head should be self-draining. Where buildings can not be retrofitted, other

approaches to minimize the growth of *Legionella* spp. include a) periodically increasing the temperature to at least 150°F [66°C] at the point of use [i.e., faucets] and b) adding additional chlorine and flushing the water.[661, 710, 711] Systems should be inspected annually to ensure that thermostats are functioning properly.

Adequate water pressure ensures sufficient water supplies for a) direct patient care; b) operation of water-cooled instruments and equipment [e.g., lasers, computer systems, telecommunications systems, and automated endoscope reprocessors[712]]; c) proper function of vacuum suctioning systems; d) indoor climate control; and e) fire-protection systems. Maintaining adequate pressure also helps to ensure the integrity of the piping system.

c. Infection-Control Impact of Water System Maintenance and Repair

Corrective measures for water-system failures have not been studied in well-designed experiments; these measures are instead based on empiric engineering and infection-control principles. Health-care facilities can occasionally sustain both intentional cut-offs by the municipal water authority to permit new construction project tie-ins and unintentional disruptions in service when a water main breaks as a result of aging infrastructure or a construction accident. Vacuum breakers or other similar devices can prevent backflow of water in the facility's distribution system during water-disruption emergencies.[11] To be prepared for such an emergency, all health-care facilities need contingency plans that identify a) the total demand for potable water, b) the quantity of replacement water [e.g., bottled water] required for a minimum of 24 hours when the water system is down, c) mechanisms for emergency water distribution, and 4) procedures for correcting drops in water pressure that affect operation of essential devices and equipment that are driven or cooled by a water system [Table 16].[713]

Table 16. Water demand in health-care facilities during water disruption emergencies

	Potable water	Bottled, sterile water
Water use needs	Drinking water Handwashing Cafeteria services Ice Manual flushing of toilets Patient baths, hygiene Hemodialysis Hydrotherapy Fire prevention (e.g., sprinkler systems) Surgery and critical care areas Laboratory services Laundry and central sterile services* Cooling towers+ Steam generation	Surgical scrub Emergency surgical procedures Pharmaceutical preparations Patient-care equipment (e.g., ventilators)§

* Arrange to have a contingency provision of these services from another resource, if possible (e.g., another health-care facility or contractor).
\+ Some cooling towers may use a potable water source, but most units use non-potable water.
§ This item is included in the table under the assumption that electrical power is available during the water emergency.

Detailed, up-to-date plans for hot and cold water piping systems should be readily available for maintenance and repair purposes in case of system problems. Opening potable water systems for repair or construction and subjecting systems to water-pressure changes can result in water discoloration and dramatic increases in the concentrations of *Legionella* spp. and other gram-negative bacteria. The maintenance of a chlorine residual at all points within the piping system also offers some protection from entry of contamination to the pipes in the event of inadvertent cross-connection between potable and non-potable water lines. As a minimum preventive measure, ASHRAE recommends a thorough flushing of the system.[661] High-temperature flushing or hyperchlorination may also be appropriate

strategies to decrease potentially high concentrations of waterborne organisms. The decision to pursue either of these remediation strategies, however, should be made on a case-by-case basis. If only a portion of the system is involved, high temperature flushing or chlorination can be used on only that portion of the system.[661]

When shock decontamination of hot water systems is necessary (e.g., after disruption caused by construction and after cross-connections), the hot water temperature should be raised to 160°F–170°F (71°C–77°C) and maintained at that level while each outlet around the system is progressively flushed. A minimum flush time of 5 minutes has been recommended;[3] the optimal flush time is not known, however, and longer flush times may be necessary.[714] The number of outlets that can be flushed simultaneously depends on the capacity of the water heater and the flow capability of the system. Appropriate safety procedures to prevent scalding are essential. When possible, flushing should be performed when the fewest building occupants are present (e.g., during nights and weekends).

When thermal shock treatment is not possible, shock chlorination may serve as an alternative method.[661] Experience with this method of decontamination is limited, however, and high levels of free chlorine can corrode metals. Chlorine should be added, preferably overnight, to achieve a free chlorine residual of at least 2 mg/L (2 ppm) throughout the system.[661] This may require chlorination of the water heater or tank to levels of 20–50 mg/L (20–50 ppm). The pH of the water should be maintained at 7.0–8.0.[661] After completion of the decontamination, recolonization of the hot water system is likely to occur unless proper temperatures are maintained or a procedure such as continuous supplemental chlorination is continued.

Interruptions of the water supply and sewage spills are situations that require immediate recovery and remediation measures to ensure the health and safety of patients and staff.[715] When delivery of potable water through the municipal distribution system has been disrupted, the public water supplier must issue a "boil water" advisory if microbial contamination presents an immediate public health risk to customers. The hospital engineer should oversee the restoration of the water system in the facility and clear it for use when appropriate. Hospitals must maintain a high level of surveillance for waterborne disease among patients and staff after the advisory is lifted.[642]

Flooding from either external (e.g., from a hurricane) or internal sources (e.g., a water system break) usually results in property damage and a temporary loss of water and sanitation.[716–718] JCAHO requires all hospitals to have plans that address facility response for recovery from both internal and external disasters.[713, 719] The plans are required to discuss a) general emergency preparedness, b) staffing, c) regional planning among area hospitals, d) emergency supply of potable water, e) infection control and medical services needs, f) climate control, and g) remediation. The basic principles of structural recovery from flooding are similar to those for recovery from sewage contamination (Box 9 and 10). Following a major event (e.g., flooding), facilities may elect to conduct microbial sampling of water after the system is restored to verify that water quality has been returned to safe levels (<500 CFU/mL, heterotrophic plate count). This approach may help identify point-of-use fixtures that may harbor contamination as a result of design or engineering features.[720] Medical records should be allowed to dry and then either photocopied or placed in plastic covers before returning them to the record.

Moisture meters can be used to assess water-damaged structural materials. If porous structural materials for walls have a moisture content of >20% after 72 hours, the affected material should be removed.[266, 278, 313] The management of water-damaged structural materials is not strictly limited to major water catastrophes (e.g., flooding and sewage intrusions); the same principles are used to evaluate the damage from leaking roofs, point-of-use fixtures, and equipment. Additional sources of moisture include condensate on walls from boilers and poorly engineered humidification in HVAC systems.

Box 9. Recovery and remediation measures for water-related emergencies*

Potable water disruptions

Contingency plan items

Ensure access to plumbing network so that repairs can be easily made.
Provide sufficient potable water, either from bottled sources or truck delivery.
Post advisory notices against consuming tap water, ice, or beverages made with water.
Rope off or bag drinking fountains to designate these as being "out of service" until further notice.
Rinse raw foods as needed in disinfected water.
Disconnect ice machines whenever possible.+
Postpone laundry services until after the water system is restored.

Water treatment

Heat water to a rolling boil for ≥1 minute.

Remediation of the water system after the "boil water" advisory is rescinded

Flush fixtures (e.g., faucets and drinking fountains) and equipment for several minutes and restart.
Run water softeners through a regeneration cycle.
Drain, disinfect, and refill water storage tanks, if needed.
Change pretreatment filters and disinfect the dialysis water system.

Sewage spills/malfunction

Overall strategy

Move patients and clean/sterile supplies out of the area.
Redirect traffic away from the area.
Close the doors or use plastic sheeting to isolate the area prior to clean-up.
Restore sewage system function first, then the potable water system (if both are malfunctioning).
Remove sewage solids, drain the area, and let dry.

Remediation of the structure

Hard surfaces: clean with detergent/disinfectant after the area has been drained.
Carpeting, loose tiles, buckled flooring: remove and allow the support surface to dry; replace the items; wet down carpeting with a low-level disinfectant or sanitizer prior to removal to minimize dust dispersion to the air.
Wallboard and other porous structural materials: remove and replace if they cannot be cleaned and dried within 72 hours.§

Furniture

Hard surface furniture (e.g., metal or plastic furniture): clean and allow to dry.
Wood furniture: let dry, sand the wood surface, and reapply varnish.
Cloth furniture: replace.

Electrical equipment

Replace if the item cannot be easily dismantled, cleaned, and reassembled.

* Material in this box is compiled from references 266, 278, 315, 713, 716–719, 721–729.
\+ Ice machines should always be disconnected from the water source in advance of planned water disruptions.
§ Moisture meter readings should be <20% moisture content.

An exception to these recommendations is made for hemodialysis units where water is further treated either by portable water treatment or large-scale water treatment systems usually involving reverse osmosis (RO). In the United States, >97% of dialysis facilities use RO treatment for their water.[721] However, changing pre-treatment filters and disinfecting the system to prevent colonization of the RO membrane and microbial contamination down-stream of the pre-treatment filter are prudent measures.

Box 10. Contingency planning for flooding

General emergency preparedness

- Ensure that emergency electrical generators are not located in flood-prone areas of the facility.
- Develop alternative strategies for moving patients, water containers, medical records, equipment, and supplies in the event that the elevators are inoperable.
- Establish in advance a centralized base of operations with batteries, flashlights, and cellular phones.
- Ensure sufficient supplies of sandbags to place at the entrances and the area around boilers, incinerators, and generators.
- Establish alternative strategies for bringing core employees to the facility if high water prevents travel.

Staffing Patterns

- Temporarily reassign licensed staff as needed to critical care areas to provide manual ventilation and to perform vital assessments on patients.
- Designate a core group of employees to remain on site to keep all services operational if the facility remains open.
- Train all employees in emergency preparedness procedures.

Regional planning among are facilities for disaster management

- Incorporate community support and involvement (e.g., media alerts, news, and transportation).
- Develop in advance strategies for transferring patients, as needed.
- Develop strategies for sharing supplies and providing essential services among participating facilities (e.g., central sterile department services, and laundry services).
- Identify sources for emergency provisions (e.g., blood, emergency vehicles, and bottled water).

Medical services and infection control

- Use alcohol-based hand rubs in general patient-care areas.
- Postpone elective surgeries until full services are restored, or transfer these patients to other facilities.
- Consider using portable dialysis machines.+
- Provide an adequate supply of tetanus and hepatitis A immunizations for patients and staff.

Climate control

- Provide adequate water for cooling towers.§

* Material in this box was compiled from references 713, 716–719.
+ Portable dialysis machines require less water compared to the larger units situated in dialysis settings.
§ Water for cooling towers may need to be trucked in, especially if the tower uses a potable water source.

4. Strategies for Controlling Waterborne Microbial Contamination

a. Supplemental Treatment of Water with Heat and/or Chemicals

In addition to using supplemental treatment methods as remediation measures after inadvertent contamination of water systems, health-care facilities sometimes use special measures to control waterborne microorganisms on a sustained basis. This decision is most often associated with outbreaks of legionellosis and subsequent efforts to control legionellae,[722] although some facilities have tried supplemental measures to better control thermophilic NTM.[627]

The primary disinfectant for both cold and hot water systems is chlorine. However, chlorine residuals are expected to be low, and possibly nonexistent, in hot water tanks because of extended retention time in the tank and elevated water temperature. Flushing, especially that which removes sludge from the bottom of the tank, probably provides the most effective treatment of water systems. Unlike the situation for disinfecting cooling towers, no equivalent recommendations have been made for potable water systems, although specific intervention strategies have been published.[403, 723] The principal approaches to disinfection of potable systems are heat flushing using temperatures 160°F–170°F (71°–77°C), hyperchlorination, and physical cleaning of hot-water tanks.[3, 403, 661] Potable systems are easily recolonized and may require continuous intervention (e.g., raising of hot water temperatures or continuous chlorination).[403, 711] Chlorine solutions lose potency over time, thereby rendering the stocking of large quantities of chlorine impractical.

Some hospitals with hot water systems identified as the source of *Legionella* spp. have performed emergency decontamination of their systems by pulse (i.e., one-time) thermal disinfection/superheating or hyperchlorination.[711, 714, 724, 725] After either of these procedures, hospitals either maintain their heated water with a minimum return temperature of 124°F (51°C) and cold water at <68°F (<20°C) or chlorinate their hot water to achieve 1–2 mg/L (1–2 ppm) of free residual chlorine at the tap.[26, 437, 709–711, 726, 727] Additional measures (e.g., physical cleaning or replacement of hot-water storage tanks, water heaters, faucets, and shower heads) may be required to help eliminate accumulations of scale and sediment that protect organisms from the biocidal effects of heat and chlorine.[457, 711] Alternative methods for controlling and eradicating legionellae in water systems (e.g., treating water with chlorine dioxide, heavy metal ions [i.e., copper/silver ions], ozone, and UV light) have limited the growth of legionellae under laboratory and operating conditions.[728–742] Further studies on the long-term efficacy of these treatments are needed before these methods can be considered standard applications.

Renewed interest in the use of chloramines stems from concerns about adverse health effects associated with disinfectants and disinfection by-products.[743] Monochloramine usage minimizes the formation of disinfection by-products, including trihalomethanes and haloacetic acids. Monochloramine can also reach distal points in a water system and can penetrate into bacterial biofilms more effectively than free chlorine.[744] However, monochloramine use is limited to municipal water treatment plants and is currently not available to health-care facilities as a supplemental water-treatment approach. A recent study indicated that 90% of Legionnaires disease outbreaks associated with drinking water could have been prevented if monochloramine rather than free chlorine has been used for residual disinfection.[745] In a retrospective comparison of health-care–associated Legionnaires disease incidence in central Texas hospitals, the same research group documented an absence of cases in facilities located in communities with monochloramine-treated municipal water.[746] Additional data are needed regarding the effectiveness of using monochloramine before its routine use as a disinfectant in water systems can be recommended. No data have been published regarding the effectiveness of monochloramine installed at the level of the health-care facility.

Additional filtration of potable water systems is not routinely necessary. Filters are used in water lines in dialysis units, however, and may be inserted into the lines for specific equipment (e.g., endoscope washers and disinfectors) for the purpose of providing bacteria-free water for instrument reprocessing. Additionally, an RO unit is usually added to the distribution system leading to PE areas.

b. Primary Prevention of Legionnaires Disease (No Cases Identified)

The primary and secondary environmental infection-control strategies described in this section on the guideline pertain to health-care facilities without transplant units. Infection-control measures specific to PE or transplant units (i.e., patient-care areas housing patients at the highest risk for morbidity and mortality from *Legionella* spp. infection) are described in the subsection titled *Preventing Legionnaires Disease in Protective Environments*.

Health-care facilities use at least two general strategies to prevent health-care–associated legionellosis when no cases or only sporadic cases have been detected. The first is an environmental surveillance approach involving periodic culturing of water samples from the hospital's potable water system to monitor for *Legionella* spp.[747–750] If any sample is culture-positive, diagnostic testing is recommended for all patients with health-care–associated pneumonia.[748, 749] In-house testing is recommended for facilities with transplant programs as part of a comprehensive treatment/management program. If $\geq$30% of the samples are culture-positive for *Legionella* spp., decontamination of the facility's potable water system is warranted.[748] The premise for this approach is that no cases of health-care–associated legionellosis can occur if *Legionella* spp. are not present in the potable water system, and, conversely, cases of health-care–associated legionellosis could potentially occur if *Legionella* spp. are cultured from the water.[26, 751] Physicians who are informed that the hospital's potable water system is culture-positive

for *Legionella* spp. are more likely to order diagnostic tests for legionellosis.

A potential advantage of the environmental surveillance approach is that periodic culturing of water is less costly than routine laboratory diagnostic testing for all patients who have health-care–associated pneumonia. The primary argument against this approach is that, in the absence of cases, the relationship between water-culture results and legionellosis risk remains undefined.[3] *Legionnella* spp. can be present in the water systems of buildings[752] without being associated with known cases of disease.[437, 707, 753] In a study of 84 hospitals in Québec, 68% of the water systems were found to be colonized with *Legionella* spp., and 26% were colonized at >30% of sites sampled; cases of Legionnaires disease, however, were infrequently reported from these hospitals.[707]

Other factors also argue against environmental surveillance. Interpretation of results from periodic water culturing might be confounded by differing results among the sites sampled in a single water system and by fluctuations in the concentration of *Legionella* spp. at the same site.[709, 754] In addition, the risk for illness after exposure to a given source might be influenced by several factors other than the presence or concentration of organisms, including a) the degree to which contaminated water is aerosolized into respirable droplets, b) the proximity of the infectious aerosol to the potential host, c) the susceptibility of the host, and d) the virulence properties of the contaminating strain.[755–757] Thus, data are insufficient to assign a level of disease risk even on the basis of the number of colony-forming units detected in samples from areas for immunocompetent patients. Conducting environmental surveillance would obligate hospital administrators to initiate water-decontamination programs if *Legionella* spp. are identified. Therefore, periodic monitoring of water from the hospital's potable water system and from aerosol-producing devices is not widely recommended in facilities that have not experienced cases of health-care–associated legionellosis.[661, 758]

The second strategy to prevent and control health-care–associated legionellosis is a clinical approach, in which providers maintain a high index of suspicion for legionellosis and order appropriate diagnostic tests (i.e., culture, urine antigen, and direct fluorescent antibody [DFA] serology) for patients with health-care–associated pneumonia who are at high risk for legionellosis and its complications.[437, 759, 760] The testing of autopsy specimens can be included in this strategy should a death resulting from health-care–associated pneumonia occur. Identification of one case of definite or two cases of possible health-care–associated Legionnaires disease should prompt an epidemiologic investigation for a hospital source of *Legionella* spp., which may involve culturing the facility's water for *Legionella*. Routine maintenance of cooling towers, and use of sterile water for the filling and terminal rinsing of nebulization devices and ventilation equipment can help to minimize potential sources of contamination. Circulating potable water temperatures should match those outlined in the subsection titled *Water Temperature and Pressure*, as permitted by state code.

c. Secondary prevention of Legionnaires Disease (With Identified Cases)

The indications for a full-scale environmental investigation to search for and subsequently decontaminate identified sources of *Legionella* spp. in health-care facilities without transplant units have not been clarified; these indications would likely differ depending on the facility. Case categories for health-care–associated Legionnaires disease in facilities without transplant units include definite cases (i.e., laboratory-confirmed cases of legionellosis that occur in patients who have been hospitalized continuously for $\geq$10 days before the onset of illness) and possible cases (i.e., laboratory-confirmed infections that occur 2–9 days after hospital admission).[3] In settings in which as few as one to three health-care–associated cases are recognized over several months, intensified surveillance for Legionnaires disease has frequently identified numerous additional cases.[405, 408, 432, 453, 739, 759, 760] This finding suggests the need for a low threshold for initiating an investigation after laboratory confirmation of cases of health-care–associated legionellosis. When developing a strategy for responding to such a finding, however, infection-control personnel should consider the level of risk for health-care–

associated acquisition of, and mortality from, *Legionella* spp. infection at their particular facility.

An epidemiologic investigation conducted to determine the source of *Legionella* spp. involves several important steps (Box 11). Laboratory assessment is crucial in supporting epidemiologic evidence of a link between human illness and a specific environmental source.[761] Strain determination from subtype analysis is most frequently used in these investigations.[410, 762–764] Once the environmental source is established and confirmed with laboratory support, supplemental water treatment strategies can be initiated as appropriate.

Box 11. Steps in an epidemiologic investigation for legionellosis

Review medical and microbiologic records.
Initiate active surveillance to identify all recent or ongoing cases.
Develop a line listing of cases by time, place, and person.
Determine the type of epidemiologic investigation needed for assessing risk factors:
- **Case-control study,**
- **Cohort study.**

Gather and analyze epidemiologic information:
- **Evaluate risk factors associated with potential environmental exposures (e.g., showers, cooling towers, and respiratory-therapy equipment).**

Collect water samples:
- **Sample environmental sources implicated by epidemiologic investigation,**
- **Sample other potential source of water aerosols.**

Subtype strains of *Legionella* spp. cultured from both patients and environmental sources.
Review autopsy records and include autopsy specimens in diagnostic testing.

The decision to search for hospital environmental sources of *Legionella* spp. and the choice of procedures to eradicate such contamination are based on several considerations, as follows: a) the hospital's patient population; b) the cost of an environmental investigation and institution of control measures to eradicate *Legionella* spp. from the water supply;[765–768] and c) the differential risk, based on host factors, for acquiring health-care–associated legionellosis and developing severe and fatal infection.

d. Preventing Legionnaires Disease in Protective Environments

This subsection outlines infection-control measures applicable to those health-care facilities providing care to severely immunocompromised patients. Indigenous microorganisms in the tap water of these facilities may pose problems for such patients. These measures are designed to prevent the generation of potentially infectious aerosols from water and the subsequent exposure of PE patients or other immunocompromised patients (e.g., transplant patients) (Table 17). Infection-control measures that address the use of water with medical equipment (e.g., ventilators, nebulizers, and equipment humidifiers) are described in other guidelines and publications.[3, 455]

If one case of laboratory-confirmed, health-care–associated Legionnaires disease is identified in a patient in a solid-organ transplant program or in PE (i.e., an inpatient in PE for all or part of the 2–10 days prior to onset of illness) or if two or more laboratory-confirmed cases occur among patients who had visited an outpatient PE setting, the hospital should report the cases to the local and state health departments. The hospital should then initiate a thorough epidemiologic and environmental investigation to determine the likely environmental sources of *Legionella* spp.[9] The source of *Legionella* should be decontaminated or removed. Isolated cases may be difficult to investigate. Because transplant recipients are at substantially higher risk for disease and death from legionellosis

compared with other hospitalized patients, periodic culturing for *Legionella* spp. in water samples from the potable water in the solid-organ transplant and/or PE unit can be performed as part of an overall strategy to prevent Legionnaires disease in PE units.[9, 431, 710, 769] The optimal methodology (i.e., frequency and number of sites) for environmental surveillance cultures in PE units has not been determined, and the cost-effectiveness of this strategy has not been evaluated. Because transplant recipients are at high risk for Legionnaires disease and because no data are available to determine a safe concentration of legionellae organisms in potable water, the goal of environmental surveillance for *Legionella* spp. should be to maintain water systems with no detectable organisms.[9, 431] Culturing for legionellae may be used to assess the effectiveness of water treatment or decontamination methods, a practice that provides benefits to both patients and health-care workers.[767, 770]

Table 17. Additional infection-control measures to prevent exposure of high-risk patients to waterborne pathogens

Measures	References
• Restrict patients from taking showers if the water is contaminated with *Legionella* spp.	• 407, 412, 654, 655, 658
• Use water that is not contaminated with *Legionella* spp. for patients' sponge baths.	• 9
• Provide sterile water for drinking, tooth brushing, or for flushing nasogastric tubes.	• 9, 412
• Perform supplemental treatment of the water for the unit.	• 732
• Consider periodic monitoring (i.e., culturing) of the unit water supply for *Legionella* spp.	• 9, 431
• Remove shower heads and faucet aerators monthly for cleaning.*	• 661
• Use a 500–600 ppm (1:100 v/v dilution) solution of sodium hypochlorite to disinfect shower heads and faucet aerators.*	• 661
• Do not use large-volume room air humidifiers that create aerosols unless these are subjected to cleaning and high-level disinfection daily and filled with distilled water.	• 3
• Eliminate water-containing bath toys.+	• 30

* These measures can be considered in settings where legionellosis cases have occurred. These measures are not generally recommended in routine patient-care setting..

\+ These items have been associated with outbreaks of *Pseudomonas*.

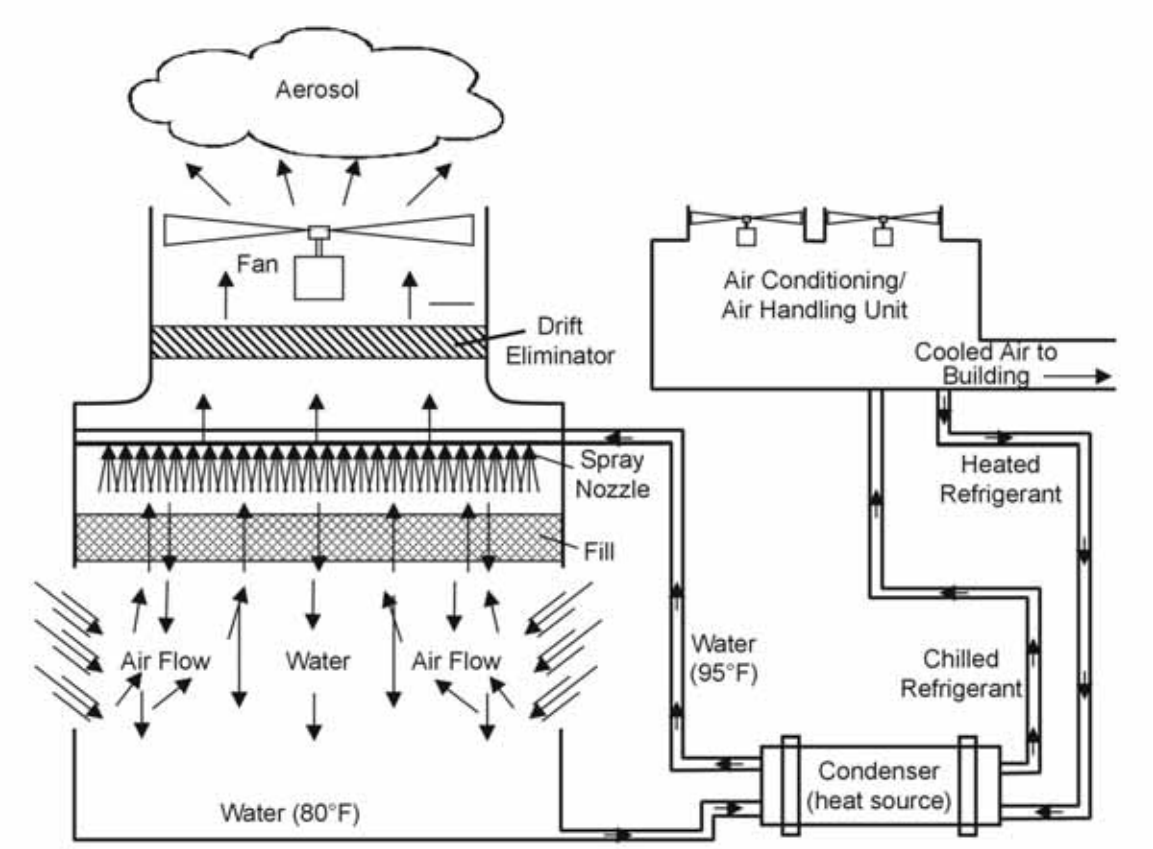
Protecting patient-care devices and instruments from inadvertent tap water contamination during room cleaning procedures is also important in any immunocompromised patient-care area. In a recent outbreak of gram-negative bacteremias among open-heart-surgery patients, pressure-monitoring equipment that was assembled and left uncovered overnight prior to the next day's surgeries was inadvertently contaminated with mists and splashing water from a hose-disinfectant system used for cleaning.[771]

5. Cooling Towers and Evaporative Condensers

Modern health-care facilities maintain indoor climate control during warm weather by use of cooling towers (large facilities) or evaporative condensers (smaller buildings). A cooling tower is a wet-type, evaporative heat transfer device used to discharge to the atmosphere waste heat from a building's air conditioning condensers (Figure 5).[772, 773] Warm water from air-conditioning condensers is piped to the cooling tower where it is sprayed downward into a counter- or cross-current air flow. To accelerate heat transfer to the air, the water passes over the fill, which either breaks water into droplets or causes it to spread into a thin film.[772, 773] Most systems use fans to move air through the tower, although some large industrial cooling towers rely on natural draft circulation of air. The cooled water from the tower is piped back to the condenser, where it again picks up heat generated during the process of chilling the system's refrigerant. The water is cycled back to the cooling tower to be cooled. Closed-circuit cooling towers and evaporative condensers are also evaporative heat-transfer devices. In these systems, the

process fluid (e.g., water, ethylene glycol/water mixture, oil, or a condensing refrigerant) does not directly contact the cooling air, but is contained inside a coil assembly.[661]

Figure 5. Diagram of a typical air conditioning (induced draft) cooling tower*



Water temperatures are approximate and may differ substantially according to system use and design. Warm water from the condenser (or chiller) is sprayed downward into a counter- or cross-current air flow. Water passes over the fill (a component of the system designed to increase the surface area of the water exposed to air), and heat from the water is transferred to the air. Some of the water becomes aerosolized during this process, although the volume of aerosol discharged to the air can be reduced by the placement of a drift eliminator. Water cooled in the tower returns to the heat source to cool refrigerant from the air conditioning unit.

* This figure is reprinted with permission of the publisher of reference 773 (Plenum Medical).

Cooling towers and evaporative condensers incorporate inertial stripping devices called drift eliminators to remove water droplets generated within the unit. Although the effectiveness of these eliminators varies substantially depending on design and condition, some water droplets in the size range of <5 µm will likely leave the unit, and some larger droplets leaving the unit may be reduced to ≤5 µm by evaporation. Thus, even with proper operation, a cooling tower or evaporative condenser can generate and expel respirable water aerosols. If either the water in the unit's basin or the make-up water (added to replace water lost to evaporation) contains *Legionella* spp. or other waterborne microorganisms, these organisms can be aerosolized and dispersed from the unit.[774] Clusters of both Legionnaires disease and Pontiac fever have been traced to exposure to infectious water aerosols originating from cooling towers and evaporative condensers contaminated with *Legionella* spp. Although most of these outbreaks have been community-acquired episodes of pneumonia,[775–782] health-care–associated Legionnaires disease

has been linked to cooling tower aerosol exposure.[404, 405] Contaminated aerosols from cooling towers on hospital premises gained entry to the buildings either through open windows or via air handling system intakes located near the tower equipment.

Cooling towers and evaporative condensers provide ideal ecological niches for *Legionella* spp. The typical temperature of the water in cooling towers ranges from 85°F–95°F (29°C–35°C), although temperatures can be above 120°F (49°C) and below 70°F (21°C) depending on system heat load, ambient temperature, and operating strategy.[661] An Australian study of cooling towers found that legionellae colonized or multiplied in towers with basin temperatures above 60.8°F (16°C), and multiplication became explosive at temperatures above 73.4°F (23°C).[783] Water temperature in closed-circuit cooling towers and evaporative condensers is similar to that in cooling towers. Considerable variation in the piping arrangement occurs. In addition, stagnant areas or dead legs may be difficult to clean or penetrate with biocides.

Several documents address the routine maintenance of cooling towers, evaporative condensers, and whirlpool spas.[661, 784–787] They suggest following manufacturer's recommendations for cleaning and biocide treatment of these devices; all health-care facilities should ensure proper maintenance for their cooling towers and evaporative condensers, even in the absence of *Legionella* spp (Appendix C). Because cooling towers and evaporative condensers can be shut down during periods when air conditioning is not needed, this maintenance cleaning and treatment should be performed before starting up the system for the first time in the warm season.[782] Emergency decontamination protocols describing cleaning procedures and hyperchlorination for cooling towers have been developed for towers implicated in the transmission of legionellosis.[786, 787]

6. Dialysis Water Quality and Dialysate

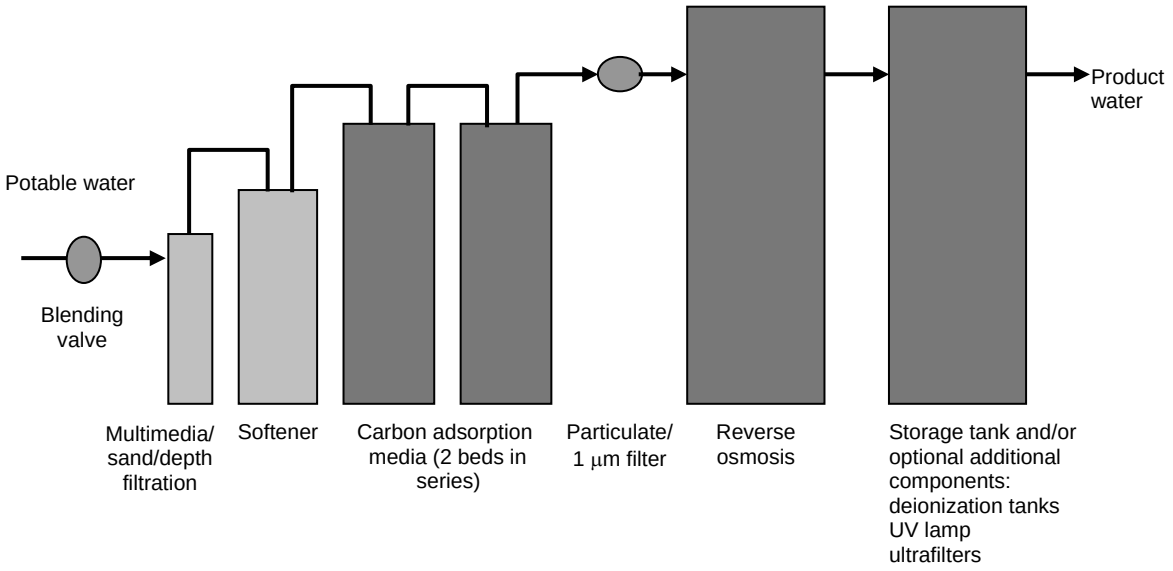
a. Rationale for Water Treatment in Hemodialysis

Hemodialysis, hemofiltration, and hemodiafiltration require special water-treatment processes to prevent adverse patient outcomes of dialysis therapy resulting from improper formulation of dialysate with water containing high levels of certain chemical or biological contaminants. The Association for the Advancement of Medical Instrumentation (AAMI) has established chemical and microbiologic standards for the water used to prepare dialysate, substitution fluid, or to reprocess hemodialyzers for renal replacement therapy.[788–792] The AAMI standards address: a) equipment and processes used to purify water for the preparation of concentrates and dialysate and the reprocessing of dialyzers for multiple use and b) the devices used to store and distribute this water. Future revisions to these standards may include hemofiltration and hemodiafiltration.

Water treatment systems used in hemodialysis employ several physical and/or chemical processes either singly or in combination (Figure 6). These systems may be portable units or large systems that feed several rooms. In the United States, >97% of maintenance hemodialysis facilities use RO alone or in combination with deionization.[793] Many acute-care facilities use portable hemodialysis machines with attached portable water treatment systems that use either deionization or RO. These machines were exempted from earlier versions of AAMI recommendations, but given current knowledge about toxic exposures to and inflammatory processes in patients new to dialysis, these machines should now come into compliance with current AAMI recommendations for hemodialysis water and dialysate quality.[788, 789] Previous recommendations were based on the assumption that acute-care patients did not experience the same degree of adverse effects from short-term, cumulative exposures to either chemicals or microbiologic agents present in hemodialysis fluids compared with the effects encountered by patients during chronic, maintenance dialysis.[788, 789] Additionally, JCAHO is reviewing inpatient

practices and record-keeping for dialysis (acute and maintenance) for adherence to AAMI standards and recommended practices.

Figure 6. Dialysis water treatment system*



* See text for description of the placement and function of these components.

Neither the water used to prepare dialysate nor the dialysate itself needs to be sterile, but tap water can not be used without additional treatment. Infections caused by rapid-growing NTM (e.g., *Mycobacterium chelonae* and *M. abscessus*) present a potential risk to hemodialysis patients (especially those in hemodialyzer reuse programs) if disinfection procedures to inactivate mycobacteria in the water (low-level disinfection) and the hemodialyzers (high-level disinfection) are inadequate.[31, 32, 633] Other factors associated with microbial contamination in dialysis systems could involve the water treatment system, the water and dialysate distribution systems, and the type of hemodialyzer.[666, 667, 794–799] Understanding the various factors and their influence on contamination levels is the key to preventing high levels of microbial contamination in dialysis therapy.

In several studies, pyrogenic reactions were demonstrated to have been caused by lipopolysaccharide or endotoxin associated with gram-negative bacteria.[794, 800–803] Early studies demonstrated that parenteral exposure to endotoxin at a concentration of 1 ng/kg body weight/hour was the threshold dose for producing pyrogenic reactions in humans, and that the relative potencies of endotoxin differ by bacterial species.[804, 805] Gram-negative water bacteria (e.g., *Pseudomonas* spp.) have been shown to multiply rapidly in a variety of hospital-associated fluids that can be used as supply water for hemodialysis (e.g., distilled water, deionized water, RO water, and softened water) and in dialysate (a balanced salt solution made with this water).[806] Several studies have demonstrated that the attack rates of pyrogenic reactions are directly associated with the number of bacteria in dialysate.[666, 667, 807] These studies provided the rationale for setting the heterotrophic bacteria standards in the first AAMI hemodialysis guideline at ≤2,000 CFU/mL in dialysate and one log lower (≤200 CFU/mL) for the water used to prepare dialysate.[668, 788] If the level of bacterial contamination exceeded 200 CFU/mL in water, this level could be amplified in the system and effectively constitute a high inoculum for dialysate at the start of a

dialysis treatment.[807, 808] Pyrogenic reactions did not appear to occur when the level of contamination was below 2,000 CFU/mL in dialysate unless the source of the endotoxin was exogenous to the dialysis system (i.e., present in the community water supply). Endotoxins in a community water supply have been linked to the development of pyrogenic reactions among dialysis patients.[794]

Whether endotoxin actually crosses the dialyzer membrane is controversial. Several investigators have shown that bacteria growing in dialysate-generated products that could cross the dialyzer membrane.[809, 810] Gram-negative bacteria growing in dialysate have produced endotoxins that in turn stimulated the production of anti-endotoxin antibodies in hemodialysis patients;[801, 811] these data suggest that bacterial endotoxins, although large molecules, cross dialyzer membranes either intact or as fragments. The use of the very permeable membranes known as high-flux membranes (which allow large molecules [e.g., β_2 microglobulin] to traverse the membrane) increases the potential for passage of endotoxins into the blood path. Several studies support this contention. In one such study, an increase in plasma endotoxin concentrations during dialysis was observed when patients were dialyzed against dialysate containing 10^3–10^4 CFU/mL *Pseudomonas* spp.[812] *In vitro* studies using both radiolabeled lipopolysaccharide and biologic assays have demonstrated that biologically active substances derived from bacteria found in dialysate can cross a variety of dialyzer membranes.[802, 813–816] Patients treated with high-flux membranes have had higher levels of anti-endotoxin antibodies than subjects or patients treated with conventional membranes.[817] Finally, since 1989, 19%–22% of dialysis centers have reported pyrogenic reactions in the absence of septicemia.[818, 819]

Investigations of adverse outcomes among patients using reprocessed dialyzers have demonstrated a greater risk for developing pyrogenic reactions when the water used to reprocess these devices contained >6 ng/mL endotoxin and $>10^4$ CFU/mL bacteria.[820] In addition to the variability in endotoxin assays, host factors also are involved in determining whether a patient will mount a response to endotoxin.[803] Outbreak investigations of pyrogenic reactions and bacteremias associated with hemodialyzer reuse have demonstrated that pyrogenic reactions are prevented once the endotoxin level in the water used to reprocess the dialyzers is returned to below the AAMI standard level.[821]

Reuse of dialyzers and use of bicarbonate dialysate, high-flux dialyzer membranes, or high-flux dialysis may increase the potential for pyrogenic reactions if the water in the dialysis setting does not meet standards.[796–798] Although investigators have been unable to demonstrate endotoxin transfer across dialyzer membranes,[803, 822, 823] the preponderance of reports now supports the ability of endotoxin to transfer across at least some high-flux membranes under some operating conditions. In addition to the acute risk of pyrogenic reactions, indirect evidence in increasingly demonstrating that chronic exposure to low amounts of endotoxin may play a role in some of the long-term complications of hemodialysis therapy. Patients treated with ultrafiltered dialysate for 5–6 months have demonstrated a decrease in serum β_2 microglobulin concentrations and a decrease in markers of an inflammatory response.[824–826] In studies of longer duration, use of microbiologically ultrapure dialysate has been associated with a decreased incidence of β_2 microglobulin-associated amyloidosis.[827, 828]

Although patient benefit likely is associated with the use of ultrapure dialysate, no consensus has been reached regarding the potential adoption of this as standard in the United States. Debate continues regarding the bacterial and endotoxin limits for dialysate. As advances in water treatment and hemodialysis processes occur, efforts are underway to move improved technology from the manufacturer out into the user community. Cost-benefit studies, however, have not been done, and substantially increased costs to implement newer water treatment modalities are anticipated.

To reconcile AAMI documents with current International Organization for Standardization (ISO) format, AAMI has determined that its hemodialysis standards will be discussed in the following four installments: RD 5 for hemodialysis equipment, RD 62 for product water quality, RD 47 for dialyzer

reprocessing, and RD 52 for dialysate quality. The Renal Diseases and Dialysis Committee of AAMI is expected to finalize and promulgated the dialysate standard pertinent to the user community (RD 52), adopting by reference the bacterial and endotoxin limits in product water as currently outlined in the AAMI standard that applies to systems manufacturers (RD 62). At present, the user community should continue to observe water quality and dialysate standards as outlined in AAMI RD 5 (Hemodialysis Systems, 1992) and AAMI RD 47 (Reuse of Hemodialyzers, 1993) until the new RD 52 standard becomes available (Table 18).[789, 791]

Table 18. Microbiologic limits for hemodialysis fluids*

Hemodialysis fluid	Maximum total heterotrophs (CFU/mL)+	Maximum endotoxin level (EU/mL)§
Present standard		
Product water¶		
Used to prepare dialysate	200	No standard
Used to reprocess dialyzers	200	5
Dialysate	2,000	No standard
Proposed standard*		
Product water	200	2
Dialysate	200	2

* The material in this table was compiled from references 789 and 791 (ANSI/AAMI standards RD 5-1992 and ANSI/AAMI RD 47-1993).
\+ Colony forming units per milliliter.
§ Endotoxin units per milliliter.
¶ Product water presently includes water used to prepare dialysate and water used to reprocess dialyzers.
** Dialysate for hemodialysis, RD 52, under development, American National Standards Institute, Association for the Advancement of Medical Instrumentation (AAMI).

The current AAMI standard directed at systems manufacturers (RD 62 [Water Treatment Equipment for Hemodialysis Applications, 2001]) now specifies that all product water used to prepare dialysate or to reprocess dialyzers for multiple use should contain <2 endotoxin units per milliliter (EU/mL).[792] A level of 2 EU/mL was chosen as the upper limit for endotoxin because this level is easily achieved with contemporary water treatment systems using RO and/or ultrafiltration. CDC has advocated monthly endotoxin testing along with microbiologic assays of water, because endotoxin activity may not correspond to the total heterotrophic plate counts.[829] Additionally, the current AAMI standard RD 62 for manufacturers includes action levels for product water. Because 48 hours can elapse between the time of sampling water for microbial contamination and the time when results are received, and because bacterial proliferation can be rapid, action levels for microbial counts and endotoxin concentrations are reported as 50 CFU/mL and 1 EU/mL, respectively, in this revision of the standard.[792] These recommendations will allow users to initiate corrective action before levels exceed the maximum levels established by the standard.

In hemodialysis, the net movement of water is from the blood to the dialysate, although within the dialyzer, local movement of water from the dialysate to the blood through the phenomenon of back-filtration may occur, particularly in dialyzers with highly permeable membranes.[830] In contrast, hemofiltration and hemodiaflltration feature infusion of large volumes of electrolyte solution (20–70 L) into the blood. Increasingly, this electrolyte solution is being prepared on-line from water and concentrate. Because of the large volumes of fluid infused, AAMI considered the necessity of setting more stringent requirements for water to be used in this application, but this organization has not yet established these because of lack of expert consensus and insufficient experience with on-line therapies in the United States. On-line hemofiltration and hemodiafiltration systems use sequential ultrafiltration as the final step in the preparation of infusion fluid. Several experts from AAMI concur that these

point-of-use ultrafiltration systems should be capable of further reducing the bacteria and endotoxin burden of solutions prepared from water meeting the requirements of the AAMI standard to a safe level for infusion.

b. Microbial Control Strategies

The strategy for controlling massive accumulations of gram-negative water bacteria and NTM in dialysis systems primarily involves preventing their growth through proper disinfection of water-treatment systems and hemodialysis machines. Gram-negative water bacteria, their associated lipopolysaccharides (bacterial endotoxins), and NTM ultimately come from the community water supply, and levels of these bacteria can be amplified depending on the water treatment system, dialysate distribution system, type of dialysis machine, and method of disinfection (Table 19).[634, 794, 831] Control strategies are designed to reduce levels of microbial contamination in water and dialysis fluid to relatively low levels but not to completely eradicate it.

Table 19. Factors influencing microbial contamination in hemodialysis systems

Factors	Comments
Water supply	
Source of community water	
Ground water	Contains endotoxin and bacteria
Surface water	Contains high levels of endotoxin and bacteria
Water treatment at the dialysis center	
None	Not recommended
Filtration	
Prefilter	Particulate filter to protect equipment; does not remove microorganisms
Absolute filter (depth or membrane filter)	Removes bacteria, however, unless the filter is changed frequently or disinfected, bacteria will accumulate and grow through the filter; acts as a significant reservoir of bacteria and endotoxin
Activated carbon filter	Removes organics and available chlorine or chloramines; acts as a significant reservoir of bacteria and endotoxin
Water treatment devices	
Deionization/ion-exchange softener	Both softeners and deionizers are significant reservoirs of bacteria and do not remove endotoxin.
Reverse osmosis (RO)	Removes bacteria and endotoxin, but must be disinfected; operates at high water pressure
Ultraviolet light	Kills some bacteria, but there is no residual; ultraviolet-resistant bacteria can develop if the unit is not properly maintained
Ultrafilter	Removes bacteria and endotoxin; operates on normal line pressure; can be positioned distal to deionizer; must be disinfected
Water and dialysate distribution system	
Distribution pipes	
Size	Oversized diameter and length decrease fluid flow and increase bacterial reservoir for both treated water and centrally-prepared dialysate.
Construction	Rough joints, dead ends, unused branches, and polyvinyl chloride (PVC) piping can act as bacterial reservoirs.
Elevation	Outlet taps should be located at the highest elevation to prevent loss of disinfectant; keep a recirculation loop in the system; flush unused ports routinely.
Storage tanks	Tanks are undesirable because they act as a reservoir for water bacteria; if tanks are present, they must be routinely scrubbed and disinfected.
Dialysis machines	
Single-pass	Disinfectant should have contact with ***all*** parts of the machine that are exposed to water or dialysis fluid.
Recirculating single-pass or recirculating (batch)	Recirculating pumps and machine design allow for massive contamination levels if not properly disinfected; overnight chemical germicide treatment is recommended.

Two components of hemodialysis water distribution systems – pipes (particularly those made of polyvinyl chloride [PVC]) and storage tanks – can serve as reservoirs of microbial contamination. Hemodialysis systems frequently use pipes that are wider and longer than are needed to handle the required flow, which slows the fluid velocity and increases both the total fluid volume and the wetted surface area of the system. Gram-negative bacteria in fluids remaining in pipes overnight multiply rapidly and colonize the wet surfaces, producing bacterial populations and endotoxin quantities in proportion to the volume and surface area. Such colonization results in formation of protective biofilm that is difficult to remove and protects the bacteria from disinfection.[832] Routine (i.e., monthly), low-level disinfection of the pipes can help to control bacterial contamination of the distribution system. Additional measures to protect pipes from contaminations include a) situating all outlet taps at equal elevation and at the highest point of the system so that the disinfectant cannot drain from pipes by gravity before adequate contact time has elapsed and b) eliminating rough joints, dead-end pipes, and unused branches and taps that can trap fluid and serve as reservoirs of bacteria capable of continuously inoculating the entire volume of the system.[800] Maintain a flow velocity of 3–5 ft/sec.

A storage tank in the distribution system greatly increases the volume of fluid and surface area available and can serve as a niche for water bacteria. Storage tanks are therefore not recommended for use in dialysis systems unless they are frequently drained and adequately disinfected, including scrubbing the sides of the tank to remove bacterial biofilm. An ultrafilter should be used distal to the storage tank.[808, 833]

Microbiologic sampling of dialysis fluids is recommended because gram-negative bacteria can proliferate rapidly in water and dialysate in hemodialysis systems; high levels of these organisms place patients at risk for pyrogenic reactions or health-care–associated infection.[667, 668, 808]

Health-care facilities are advised to sample dialysis fluids at least monthly using standard microbiologic assay methods for waterborne microorganisms.[788, 793, 799, 834–836] Product water used to prepare dialysate and to reprocess hemodialyzers for reuse on the same patient should also be tested for bacterial endotoxin on a monthly basis.[792, 829, 837] (See Appendix C for information about water sampling methods for dialysis.)

Cross-contamination of dialysis machines and inadequate disinfection measures can facilitate the spread of waterborne organisms to patients. Steps should be taken to ensure that dialysis equipment is performing correctly and that all connectors, lines, and other components are specific for the equipment, in good repair, and properly in place. A recent outbreak of gram-negative bacteremias among dialysis patients was attributed to faulty valves in a drain port of the machine that allowed backflow of saline used to flush the dialyzer before patient use.[838, 839] This backflow contaminated the drain priming connectors, which contaminated the blood lines and exposed the patients to high concentrations of gram-negative bacteria. Environmental infection control in dialysis settings also includes low-level disinfection of housekeeping surfaces and spot decontamination of spills of blood (see Environmental Services in Part I of this guideline for further information).

c. Infection-Control Issues in Peritoneal Dialysis

Peritoneal dialysis (PD), most commonly administered as continuous ambulatory peritoneal dialysis (CAPD) and continual cycling peritoneal dialysis (CCPD), is the third most common treatment for end-stage renal disease (ESRD) in the United States, accounting for 12% of all dialysis patients.[840] Peritonitis is the primary complication of CAPD, with coagulase-negative staphylococci the most clinically significant causative organisms.[841] Other organisms that have been found to produce peritonitis include *Staphylococcus aureus, Mycobacterium fortuitum, M. mucogenicum, Stenotrophomonas maltophilia, Burkholderia cepacia, Corynebacterium jeikeium, Candida* spp., and

other fungi.[842–850] Substantial morbidity is associated with peritoneal dialysis infections. Removal of peritoneal dialysis catheters usually is required for treatment of peritonitis caused by fungi, NTM, or other bacteria that are not cleared within the first several days of effective antimicrobial treatment. Furthermore, recurrent episodes of peritonitis may lead to fibrosis and loss of the dialysis membrane.

Many reported episodes of peritonitis are associated with exit-site or tunneled catheter infections. Risk factors for the development of peritonitis in PD patients include a) under dialysis, b) immune suppression, c) prolonged antimicrobial treatment, d) patient age [more infections occur in younger patients and older hospitalized patients], e) length of hospital stay, and f) hypoalbuminemia.[844, 851, 852] Concern has been raised about infection risk associated with the use of automated cyclers in both inpatient and outpatient settings; however, studies suggest that PD patients who use automated cyclers have much lower infection rates.[853] One study noted that a closed-drainage system reduced the incidence of system-related peritonitis among intermittent peritoneal dialysis (IPD) patients from 3.6 to 1.5 cases/100 patient days.[854] The association of peritonitis with management of spent dialysate fluids requires additional study. Therefore, ensuring that the tip of the waste line is not submerged beneath the water level in a toilet or in a drain is prudent.

7. Ice Machines and Ice

Microorganisms may be present in ice, ice-storage chests, and ice-making machines. The two main sources of microorganisms in ice are the potable water from which it is made and a transferral of organisms from hands (Table 20). Ice from contaminated ice machines has been associated with patient colonization, blood stream infections, pulmonary and gastrointestinal illnesses, and pseudoinfections.[602, 603, 683, 684, 854, 855] Microorganisms in ice can secondarily contaminate clinical specimens and medical solutions that require cold temperatures for either transport or holding.[601, 620] An outbreak of surgical-site infections was interrupted when sterile ice was used in place of tap water ice to cool cardioplegia solutions.[601]

Table 20. Microorganisms and their sources in ice and ice machines

Sources of microorganisms	References
From potable water	
Legionella spp.	684, 685, 857, 858
Nontuberculous mycobacteria (NTM)	602, 603, 859
Pseudomonas aeruginosa	859
Burkholderia cepacia	859, 860
Stenotrophomonas maltophilia	860
Flavobacterium spp.	860
From fecally-contaminated water	
Norwalk virus	861–863
Giardia lamblia	864
Cryptosporidium parvum	685
From hand-transfer of organisms	
Acinetobacter spp.	859
Coagulase-negative staphylococci	859
Salmonella enteriditis	865
Cryptosporidium parvum	685

In a study comparing the microbial populations of hospital ice machines with organisms recovered from ice samples gathered from the community, samples from 27 hospital ice machines yielded low numbers (<10 CFU/mL) of several potentially opportunistic microorganisms, mainly gram-negative bacilli.[859] During the survey period, no health-care–associated infections were attributed to the use of ice. Ice from community sources had higher levels of microbial contamination (75%–95% of 194 samples had total heterotrophic plate counts <500 CFU/mL, with the proportion of positive cultures dependent on the incubation temperature) and showed evidence of fecal contamination from the source water.[859] Thus, ice machines in health-care settings are no more heavily contaminated compared with ice machines in the community. If the source water for ice in a health-care facility is not fecally contaminated, then ice from clean ice machines and chests should pose no increased hazard for immunocompetent patients. Some waterborne bacteria found in ice could potentially be a risk to immunocompromised patients if they consume ice or drink beverages with ice. For example, *Burkholderia cepacia* in ice could present an infection risk for cystic fibrosis patients.[859, 860] Therefore, protecting immunosuppressed and otherwise medically at-risk patients from exposure to tap water and ice potentially contaminated with opportunistic pathogens is prudent.[9]

No microbiologic standards for ice, ice-making machines, or ice storage equipment have been established, although several investigators have suggested the need for such standards.[859, 866] Culturing of ice machines is not routinely recommended, but it may be useful as part of an epidemiologic investigation.[867–869] Sampling might also help determine the best schedule for cleaning open ice-storage chests. Recommendations for a regular program of maintenance and disinfection have been published.[866–869] Health-care facilities are advised to clean ice-storage chests on a regular basis. Open ice chests may require a more frequent cleaning schedule compared with chests that have covers. Portable ice chests and containers require cleaning and low-level disinfection before the addition of ice intended for consumption. Ice-making machines may require less frequent cleaning, but their maintenance is important to proper performance. The manufacturer's instructions for both the proper method of cleaning and/or maintenance should be followed. These instructions may also recommend an EPA-registered disinfectant to ensure chemical potency, materials compatibility, and safety. In the event that instructions and suitable EPA-registered disinfectants are not available for this process, then a generic approach to cleaning, disinfecting, and maintaining ice machines and dispensers can be used (Box 12).

Ice and ice-making machines also may be contaminated via improper storage or handling of ice by patients and/or staff.[684–686, 855–858, 870] Suggested steps to avoid this means of contamination include a) minimizing or avoiding direct hand contact with ice intended for consumption, b) using a hard-surface scoop to dispense ice, and c) installing machines that dispense ice directly into portable containers at the touch of a control.[687, 869]

Box 12. General steps for cleaning and maintaining ice machines, dispensers, and storage chests*+

1. **Disconnect unit from power supply.**
2. **Remove and discard ice from bin or storage chest.**
3. **Allow unit to warm to room temperature.**
4. **Disassemble removable parts of machine that make contact with water to make ice.**
5. **Thoroughly clean machine and parts with water and detergent.**
6. **Dry external surfaces of removable parts before reassembling.**
7. **Check for any needed repair.**
8. **Replace feeder lines, as appropriate (e.g., when damaged, old, or difficult to clean).**
9. **Ensure presence of an air space in tubing leading from water inlet into water distribution system of machine.**

(Box 12. continued)

10. **Inspect for rodent or insect infestations under the unit and treat, as needed.**
11. **Check door gaskets (open compartment models) for evidence of leakage or dripping into the storage chest.**
12. **Clean the ice-storage chest or bin with fresh water and detergent; rinse with fresh tap water.**
13. **Sanitize machine by circulating a 50–100 parts per million (ppm) solution of sodium hypochlorite (i.e., 4–8 mL sodium hypochlorite/gallon of water) through the ice-making and storage systems for 2 hours (100 ppm solution), or 4 hours (50 ppm solution).**
14. **Drain sodium hypochlorite solutions and flush with fresh tap water.**
15. **Allow all surfaces of equipment to dry before returning to service.**

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* Material in this box is adapted from reference 869.
+ These general guidelines should be used only where manufacturer-recommended methods and EPA-registered disinfectants are not available.

8. Hydrotherapy Tanks and Pools

a. General Information

Hydrotherapy equipment (e.g., pools, whirlpools, whirlpool spas, hot tubs, and physiotherapy tanks) traditionally has been used to treat patients with certain medical conditions (e.g., burns,[871, 872] septic ulcers, lesions, amputations,[873] orthopedic impairments and injuries, arthritis,[874] and kidney lithotripsy).[654] Wound-care medicine is increasingly moving away from hydrotherapy, however, in favor of bedside pulsed-lavage therapy using sterile solutions for cleaning and irrigation.[492, 875–878] Several episodes of health-care–associated infections have been linked to use of hydrotherapy equipment (Table 21). Potential routes of infection include incidental ingestion of the water, sprays and aerosols, and direct contact with wounds and intact skin (folliculitis). Risk factors for infection include a) age and sex of the patient, b) underlying medical conditions, c) length of time spent in the hydrotherapy water, and d) portals of entry.[879]

Table 21. Infections associated with use of hydrotherapy equipment

Microorganisms	Medical conditions	References
Acinetobacter baumanii	Sepsis	572
Citrobacter freundii	Cellulitis	880
Enterobacter cloacae	Sepsis	881
Legionella spp.	Legionellosis	882
Mycobacterium abscessus, Mycobacterium fortuitum, Mycobacterium marinum	Skin ulcers and soft tissue infections	621–623, 883
Pseudomonas aeruginosa	Sepsis, soft tissue infections, folliculitis, and wound infections	492, 493, 506, 679, 884–888
Adenovirus, adeno-associated virus	Conjunctivitis	889

Infection control for hydrotherapy tanks, pools, or birthing tanks presents unique challenges because indigenous microorganisms are always present in the water during treatments. In addition, some studies have found free living amoebae (i.e., *Naegleria lovaniensis*), which are commonly found in association with *Naegleria fowleri,* in hospital hydrotherapy pools.[890] Although hydrotherapy is at times appropriate for patients with wounds, burns, or other types of non-intact skin conditions (determined on a case-by-case basis), this equipment should not be considered "semi-critical" in accordance with the Spaulding classification.[891] Microbial data to evaluate the risk of infection to patients using hydrotherapy pools and birthing tanks are insufficient. Nevertheless, health-care facilities should maintain stringent cleaning and disinfection practices in accordance with the manufacturer's instructions

and with relevant scientific literature until data supporting more rigorous infection-control measures become available. Factors that should be considered in therapy decisions in this situation would include a) availability of alternative aseptic techniques for wound management and b) a risk-benefit analysis of using traditional hydrotherapy.

b. Hydrotherapy Tanks

Hydrotherapy tanks (e.g., whirlpools, Hubbard tanks and whirlpool bath tubs) are shallow tanks constructed of stainless steel, plexiglass, or tile. They are closed-cycle water systems with hydrojets to circulate, aerate, and agitate the water. The maximum water temperature range is 50°F–104°F (10°C–40°C). The warm water temperature, constant agitation and aeration, and design of the hydrotherapy tanks provide ideal conditions for bacterial proliferation if the equipment is not properly maintained, cleaned, and disinfected. The design of the hydrotherapy equipment should be evaluated for potential infection-control problems that can be associated with inaccessible surfaces that can be difficult to clean and/or remain wet in between uses (i.e., recessed drain plates with fixed grill plates).[887] Associated equipment (e.g., parallel bars, plinths, Hoyer lifts, and wheelchairs) can also be potential reservoirs of microorganisms, depending on the materials used in these items (i.e., porous vs. non-porous materials) and the surfaces that may become wet during use. Patients with active skin colonizations and wound infections can serve as sources of contamination for the equipment and the water. Contamination from spilled tub water can extend to drains, floors, and walls.[680–683] Health-care–associated colonization or infection can result from exposure to endogenous sources of microorganisms (autoinoculation) or exogenous sources (via cross-contamination from other patients previously receiving treatment in the unit).

Although some facilities have used tub liners to minimize environmental contamination of the tanks, the use of a tub liner does not eliminate the need for cleaning and disinfection. Draining these small pools and tanks after each patient use, thoroughly cleaning with a detergent, and disinfecting according to manufacturers' instructions have reduced bacterial contamination levels in the water from 10^4 CFU/mL to <10 CFU/mL.[892] A chlorine residual of 15 ppm in the water should be obtained prior to the patient's therapy session (e.g., by adding 15 grams of calcium hypochlorite 70% [e.g., HTH®] per 100 gallons of water).[892] A study of commercial and residential whirlpools found that superchlorination or draining, cleaning, disinfection, and refilling of whirlpools markedly reduced densities of *Pseudomonas aeruginosa* in whirlpool water.[893] The bacterial populations were rapidly replenished, however, when disinfectant concentrations dropped below recommended levels for recreational use (i.e., chlorine at 3.0 ppm or bromine at 6.0 ppm). When using chlorine, however, knowing whether the community drinking-water system is disinfected with chloramine is important, because municipal utilities adjust the pH of the water to the basic side to enhance chloramine formation. Because chlorine is not very effective at pH levels above 8, it may be necessary to re-adjust the pH of the water to a more acidic level.[894]

A few reports describe the addition of antiseptic chemicals to hydrotherapy tank water, especially for burn patient therapy.[895–897] One study involving a minimal number of participants demonstrated a reduction in the number of *Pseudomonas* spp. and other gram-negative bacteria from both patients and equipment surfaces when chloramine-T ("chlorazene") was added to the water.[898] Chloramine-T has not, however, been approved for water treatment in the United States.

c. Hydrotherapy Pools

Hydrotherapy pools typically serve large numbers of patients and are usually heated to 91.4°F–98.6°F (31°C–37°C). The temperature range is more narrow (94°F–96.8°F [35°C–36°C]) for pediatric and geriatric patient use.[899] Because the size of hydrotherapy pools precludes draining after patient use, proper management is required to maintain the proper balance of water conditioning (i.e., alkalinity, hardness, and temperature) and disinfection. The most widely used chemicals for disinfection of pools

are chlorine and chlorine compounds – calcium hypochlorite, sodium hypochlorite, lithium hypochlorite, chloroisocyanurates, and chlorine gas. Solid and liquid formulations of chlorine chemicals are the easiest and safest to use.[900] Other halogenated compounds have also been used for pool-water disinfection, albeit on a limited scale. Bromine, which forms bactericidal bromamines in the presence of ammonia, has limited use because of its association with contact dermatitis.[901] Iodine does not bleach hair, swim suits, or cause eye irritation, but when introduced at proper concentrations, it gives water a greenish-yellowish cast.[892]

In practical terms, maintenance of large hydrotherapy pools (e.g., those used for exercise) is similar to that for indoor public pools (i.e., continuous filtration, chlorine residuals no less than 0.4 ppm, and pH of 7.2–7.6).[902, 903] Supply pipes and pumps also need to be maintained to eliminate the possibility of this equipment serving as a reservoir for waterborne organisms.[904] Specific standards for chlorine residual and pH of the water are addressed in local and state regulations. Patients who are fecally incontinent or who have draining wounds should refrain from using these pools until their condition improves.

d. Birthing Tanks and Other Equipment

The use of birthing tanks, whirlpool spas, and whirlpools is a recent addition to obstetrical practice.[905] Few studies on the potential risks associated with these pieces of equipment have been conducted. In one study of 32 women, a newborn contracted a *Pseudomonas* infection after being birthed in such a tank, the strain of which was identical to the organism isolated from the tank water.[906] Another report documented identical strains of *P. aeruginosa* isolates from a newborn with sepsis and on the environmental surfaces of a tub that the mother used for relaxation while in labor.[907] Other studies have shown no significant increases in the rates of post-immersion infections among mothers and infants.[908, 909]

Because the water and the tub surfaces routinely become contaminated with the mother's skin flora and blood during labor and delivery, birthing tanks and other tub equipment must be drained after each patient use and the surfaces thoroughly cleaned and disinfected. Health-care facilities are advised to follow the manufacturer's instructions for selection of disinfection method and chemical germicide. The range of chlorine residuals for public whirlpools and whirlpool spas is 2–5 ppm.[910] Use of an inflatable tub is an alternative solution, but this item must be cleaned and disinfected between patients if it is not considered a single-use unit.

Recreational tanks and whirlpool spas are increasingly being used as hydrotherapy equipment. Although such home equipment appears to be suitable for hydrotherapy, they are neither designed nor constructed to function in this capacity. Additionally, manufacturers generally are not obligated to provide the health-care facility with cleaning and disinfecting instructions appropriate for medical equipment use, and the U.S. Food and Drug Administration (FDA) does not evaluate recreational equipment. Health-care facilities should therefore carefully evaluate this "off-label" use of home equipment before proceeding with a purchase.

9. Miscellaneous Medical/Dental Equipment Connected to Main Water Systems

a. Automated Endoscope Reprocessors

The automated endoscopic reprocessor (AER) is classified by the FDA as an accessory for the flexible endoscope.[654] A properly operating AER can provide a more consistent, reliable method of decontaminating and terminal reprocessing for endoscopes between patient procedures than manual reprocessing methods alone.[911] An endoscope is generally subjected to high-level disinfection using a

liquid chemical sterilant or a high-level disinfectant. Because the instrument is a semi-critical device, the optimal rinse fluid for a disinfected endoscope would be sterile water.[3] Sterile water, however, is expensive and difficult to produce in sufficient quantities and with adequate quality assurance for instrument rinsing in an AER.[912, 913] Therefore, one option to be used for AERs is rinse water that has been passed through filters with a pore size of 0.1–0.2 µm to render the water "bacteria-free." These filters usually are located in the water line at or near the port where the mains water enters the equipment. The product water (i.e., tap water passing through these filters) in these applications is not considered equivalent in microbial quality to that for membrane-filtered water as produced by pharmaceutical firms. Membrane filtration in pharmaceutical applications is intended to ensure the microbial quality of polished product water.

Water has been linked to the contamination of flexible fiberoptic endoscopes in the following two scenarios: a) rinsing a disinfected endoscope with unfiltered tap water, followed by storage of the instrument without drying out the internal channels and b) contamination of AERs from tap water inadvertently introduced into the equipment. In the latter instance, the machine's water reservoirs and fluid circuitry become contaminated with waterborne, heterotrophic bacteria (e.g., *Pseudomonas aeruginosa* and NTM), which can survive and persist in biofilms attached to these components.[914–917] Colonization of the reservoirs and water lines of the AER becomes problematic if the required cleaning, disinfection, and maintenance are not performed on the equipment as recommended by the manufacturer.[669, 916, 917] Use of the 0.1–0.2-µm filter in the water line helps to keep bacterial contamination to a minimum,[670, 911, 917] but filters may fail and allow bacteria to pass through to the equipment and then to the instrument undergoing reprocessing.[671–674, 913, 918] Filters also require maintenance for proper performance.[670, 911, 912, 918, 919] Heightened awareness of the proper disinfection of the connectors that hook the instrument to the AER may help to further reduce the potential for contaminating endoscopes during reprocessing.[920] An emerging issue in the field of endoscopy is that of the possible role of rinse water monitoring and its potential to help reduce endoscopy/bronchoscopy-associated infections.[918]

Studies have linked deficiencies in endoscope cleaning and/or disinfecting processes to the incidence of post-endoscopic adverse outcomes.[921–924] Several clusters have been traced to AERs of older designs and these were associated with water quality.[675, 914–916] Regardless of whether manual or automated terminal reprocessing is used for endoscopes, the internal channels of the instrument should be dried before storage.[925] The presence of residual moisture in the internal channels encourages the proliferation of waterborne microorganisms, some of which may be pathogenic. One of the most frequently used methods employs 70% isopropyl alcohol to flush the internal channels, followed by forced air drying of these channels and hanging the endoscope vertically in a protected cabinet; this method ensures internal drying of the endoscope, lessens the potential for proliferation of waterborne microorganisms,[669, 913, 917, 922, 926, 927] and is consistent with professional organization guidance for endoscope reprocessing.[928]

An additional problem with waterborne microbial contamination of AERs centers on increased microbial resistance to alkaline glutaraldehyde, a widely used liquid chemical sterilant/high-level disinfectant.[669, 929] Opportunistic waterborne microorganisms (e.g., *Mycobacterium chelonae*, *Methylobacterium* spp.) have been associated with pseudo-outbreaks and colonization; infection caused by these organisms has been associated with procedures conducted in clinical settings (e.g., bronchoscopy).[669, 913, 929–931] Increasing microbial resistance to glutaraldehyde has been attributed to improper use of the disinfectant in the equipment, allowing the dilution of glutaraldehyde to fall below the manufacturer's recommended minimal use concentration.[929]

b. Dental Unit Water Lines

Dental unit water lines (DUWLs) consist of small-bore plastic tubing that delivers water used for general, non-surgical irrigation and as a coolant to dental handpieces, sonic and ultrasonic scalers, and air-water syringes; municipal tap water is the source water for these lines. The presence of biofilms of waterborne bacteria and fungi (e.g., *Legionella* spp., *Pseudomonas aeruginosa,* and NTM) in DUWLs has been established.[636, 637, 694, 695, 932– 934] Biofilms continually release planktonic microorganisms into the water, the titers of which can exceed 1×10^6 CFU/mL.[694] However, scientific evidence indicates that immunocompetent persons are only at minimal risk for substantial adverse health effects after contact with water from a dental unit. Nonetheless, exposing patients or dental personnel to water of uncertain microbiological quality is not consistent with universally accepted infection-control principles.[935]

In 1993, CDC issued guidelines relative to water quality in a dental setting. These guidelines recommend that all dental instruments that use water (including high-speed handpieces) should be run to discharge water for 20–30 seconds after each patient and for several minutes before the start of each clinic day.[936] This practice can help to flush out any patient materials that many have entered the turbine, air, or waterlines.[937, 938] The 1993 guidance also indicated that waterlines be flushed at the beginning of the clinic day. Although these guidelines are designed to help reduce the number of microorganisms present in treatment water, they do not address the issue of reducing or preventing biofilm formation in the waterlines. Research published subsequent to the 1993 dental infection control guideline suggests that flushing the lines at the beginning of the day has only minimal effect on the status of the biofilm in the lines and does not reliably improve the quality of water during dental treatment.[939–941] Updated recommendations on infection-control practices for water line use in dentistry will be available in late 2003.[942]

The numbers of microorganisms in water used as coolant or irrigant for non-surgical dental treatment should be as low as reasonably achievable and, at a minimum, should meet nationally recognized standards for safe drinking water.[935, 943] Only minimal evidence suggests that water meeting drinking water standards poses a health hazard for immunocompetent persons. The EPA, the American Public Health Association (APHA), and the American Water Works Association (AWWA) have set a maximum limit of 500 CFU/mL for aerobic, heterotrophic, mesophilic bacteria in drinking water in municipal distribution systems.[944, 945] This standard is achievable, given improvements in water-line technology. Dentists should consult with the manufacturer of their dental unit to determine the best equipment and method for maintaining and monitoring good water quality.[935, 946]

E. Environmental Services

1. Principles of Cleaning and Disinfecting Environmental Surfaces

Although microbiologically contaminated surfaces can serve as reservoirs of potential pathogens, these surfaces generally are not directly associated with transmission of infections to either staff or patients. The transferral of microorganisms from environmental surfaces to patients is largely via hand contact with the surface.[947, 948] Although hand hygiene is important to minimize the impact of this transfer, cleaning and disinfecting environmental surfaces as appropriate is fundamental in reducing their potential contribution to the incidence of healthcare-associated infections.

The principles of cleaning and disinfecting environmental surfaces take into account the intended use of the surface or item in patient care. CDC retains the Spaulding classification for medical and surgical instruments, which outlines three categories based on the potential for the instrument to transmit infection if the instrument is microbiologically contaminated before use.[949, 950] These categories are

"critical," "semicritical," and "noncritical." In 1991, CDC proposed an additional category designated "environmental surfaces" to Spaulding's original classification[951] to represent surfaces that generally do not come into direct contact with patients during care. Environmental surfaces carry the least risk of disease transmission and can be safely decontaminated using less rigorous methods than those used on medical instruments and devices. Environmental surfaces can be further divided into medical equipment surfaces (e.g., knobs or handles on hemodialysis machines, x-ray machines, instrument carts, and dental units) and housekeeping surfaces (e.g., floors, walls, and tabletops).[951]

The following factors influence the choice of disinfection procedure for environmental surfaces: a) the nature of the item to be disinfected, b) the number of microorganisms present, c) the innate resistance of those microorganisms to the inactivating effects of the germicide, d) the amount of organic soil present, e) the type and concentration of germicide used, f) duration and temperature of germicide contact, and g) if using a proprietary product, other specific indications and directions for use.[952, 953]

Cleaning is the necessary first step of any sterilization or disinfection process. Cleaning is a form of decontamination that renders the environmental surface safe to handle or use by removing organic matter, salts, and visible soils, all of which interfere with microbial inactivation.[954–960] The physical action of scrubbing with detergents and surfactants and rinsing with water removes large numbers of microorganisms from surfaces.[957] If the surface is not cleaned before the terminal reprocessing procedures are started, the success of the sterilization or disinfection process is compromised.

Spaulding proposed three levels of disinfection for the treatment of devices and surfaces that do not require sterility for safe use. These disinfection levels are "high-level," "intermediate-level," and "low-level."[949, 950] The basis for these levels is that microorganisms can usually be grouped according to their innate resistance to a spectrum of physical or chemical germicidal agents (Table 22). This information, coupled with the instrument/surface classification, determines the appropriate level of terminal disinfection for an instrument or surface.

Table 22. Levels of disinfection by type of microorganism*

	Bacteria			Fungi+	Viruses	
Disinfection level	**Vegetative**	**Tubercle bacillus**	**Spores**		**Lipid and medium size**	**Nonlipid and small size**
High	+§	+	+¶	+	+	+
Intermediate	+	+	–**	+	+	±++
Low	+	–	–	±	+	±

* Material in this table compiled from references 2 and 951.
\+ This class of microorganisms includes asexual spores but not necessarily chlamydospores or sexual spores.
§ The "plus" sign indicates that a killing effect can be expected when the normal use-concentrations of chemical disinfectants or pasteurization are properly employed; a "negative" sign indicates little or no killing effect.
¶ Only with extended exposure times are high-level disinfectant chemicals capable of killing high numbers of bacterial spores in laboratory tests; they are, however, capable of sporicidal activity.
** Some intermediate-level disinfectants (e.g., hypochlorites) can exhibit some sporicidal activity; others (e.g., alcohols and phenolics) have no demonstrable sporicidal activity.
++ Some intermediate-level disinfectants, although they are tuberculocidal, may have limited virucidal activity.

The process of high-level disinfection, an appropriate standard of treatment for heat-sensitive, semi-critical medical instruments (e.g., flexible, fiberoptic endoscopes), inactivates all vegetative bacteria, mycobacteria, viruses, fungi, and some bacterial spores. High-level disinfection is accomplished with powerful, sporicidal chemicals (e.g., glutaraldehyde, peracetic acid, and hydrogen peroxide) that are not appropriate for use on housekeeping surfaces. These liquid chemical sterilants/high-level disinfectants

are highly toxic.[961–963] Use of these chemicals for applications other than those indicated in their label instructions (i.e., as immersion chemicals for treating heat-sensitive medical instruments) is not appropriate.[964] Intermediate-level disinfection does not necessarily kill bacterial spores, but it does inactivate *Mycobacterium tuberculosis* var. *bovis*, which is substantially more resistant to chemical germicides than ordinary vegetative bacteria, fungi, and medium to small viruses (with or without lipid envelopes). Chemical germicides with sufficient potency to achieve intermediate-level disinfection include chlorine-containing compounds (e.g., sodium hypochlorite), alcohols, some phenolics, and some iodophors. Low-level disinfection inactivates vegetative bacteria, fungi, enveloped viruses (e.g., human immunodeficiency virus [HIV], and influenza viruses), and some non-enveloped viruses (e.g., adenoviruses). Low-level disinfectants include quaternary ammonium compounds, some phenolics, and some iodophors. Sanitizers are agents that reduce the numbers of bacterial contaminants to safe levels as judged by public health requirements, and are used in cleaning operations, particularly in food service and dairy applications. Germicidal chemicals that have been approved by FDA as skin antiseptics are not appropriate for use as environmental surface disinfectants.[951]

The selection and use of chemical germicides are largely matters of judgment, guided by product label instructions, information, and regulations. Liquid sterilant chemicals and high-level disinfectants intended for use on critical and semi-critical medical/dental devices and instruments are regulated exclusively by the FDA as a result of recent memoranda of understanding between FDA and the EPA that delineates agency authority for chemical germicide regulation.[965, 966] Environmental surface germicides (i.e., primarily intermediate- and low-level disinfectants) are regulated by the EPA and labeled with EPA registration numbers. The labels and technical data or product literature of these germicides specify indications for product use and provide claims for the range of antimicrobial activity. The EPA requires certain pre-registration laboratory potency tests for these products to support product label claims. EPA verifies (through laboratory testing) manufacturers' claims to inactivate microorganisms for selected products and organisms. Germicides labeled as "hospital disinfectant" have passed the potency tests for activity against three representative microorganisms – *Pseudomonas aeruginosa, Staphylococcus aureus,* and *Salmonella cholerae suis*. Low-level disinfectants are often labeled "hospital disinfectant" without a tuberculocidal claim, because they lack the potency to inactivate mycobacteria. Hospital disinfectants with demonstrated potency against mycobacteria (i.e., intermediate-level disinfectants) may list "tuberculocidal" on the label as well. Other claims (e.g., "fungicidal," "pseudomonicidal," and "virucidal") may appear on labels of environmental surface germicides, but the designations of "tuberculocidal hospital disinfectant" and "hospital disinfectant" correlate directly to Spaulding's assessment of intermediate-level disinfectants and low-level disinfectants, respectively.[951]

A common misconception in the use of surface disinfectants in health-care settings relates to the underlying purpose for use of proprietary products labeled as a "tuberculocidal" germicide. Such products will not interrupt and prevent the transmission of TB in health-care settings because TB is not acquired from environmental surfaces. The tuberculocidal claim is used as a benchmark by which to measure germicidal potency. Because mycobacteria have the highest intrinsic level of resistance among the vegetative bacteria, viruses, and fungi, any germicide with a tuberculocidal claim on the label (i.e., an intermediate-level disinfectant) is considered capable of inactivating a broad spectrum of pathogens, including much less resistant organisms such the bloodborne pathogens (e.g., hepatitis B virus [HBV], hepatitis C virus [HCV], and HIV). It is this broad spectrum capability, rather than the product's specific potency against mycobacteria, that is the basis for protocols and OSHA regulations indicating the appropriateness of using tuberculocidal chemicals for surface disinfection.[967]

2. General Cleaning Strategies for Patient-Care Areas

The number and types of microorganisms present on environmental surfaces are influenced by the following factors: a) number of people in the environment, b) amount of activity, c) amount of moisture, d) presence of material capable of supporting microbial growth, e) rate at which organisms suspended in the air are removed, and f) type of surface and orientation [i.e., horizontal or vertical].[968] Strategies for cleaning and disinfecting surfaces in patient-care areas take into account a) potential for direct patient contact, b) degree and frequency of hand contact, and c) potential contamination of the surface with body substances or environmental sources of microorganisms (e.g., soil, dust, and water).

a. Cleaning of Medical Equipment

Manufacturers of medical equipment should provide care and maintenance instructions specific to their equipment. These instructions should include information about a) the equipments' compatibility with chemical germicides, b) whether the equipment is water-resistant or can be safely immersed for cleaning, and c) how the equipment should be decontaminated if servicing is required.[967] In the absence of manufacturers' instructions, non-critical medical equipment (e.g., stethoscopes, blood pressure cuffs, dialysis machines, and equipment knobs and controls) usually only require cleansing followed by low- to intermediate-level disinfection, depending on the nature and degree of contamination. Ethyl alcohol or isopropyl alcohol in concentrations of 60%–90% (v/v) is often used to disinfect small surfaces (e.g., rubber stoppers of multiple-dose medication vials, and thermometers)[952, 969] and occasionally external surfaces of equipment (e.g., stethoscopes and ventilators). However, alcohol evaporates rapidly, which makes extended contact times difficult to achieve unless items are immersed, a factor that precludes its practical use as a large-surface disinfectant.[951] Alcohol may cause discoloration, swelling, hardening, and cracking of rubber and certain plastics after prolonged and repeated use and may damage the shellac mounting of lenses in medical equipment.[970]

Barrier protection of surfaces and equipment is useful, especially if these surfaces are a) touched frequently by gloved hands during the delivery of patient care, b) likely to become contaminated with body substances, or c) difficult to clean. Impervious-backed paper, aluminum foil, and plastic or fluid-resistant covers are suitable for use as barrier protection. An example of this approach is the use of plastic wrapping to cover the handle of the operatory light in dental-care settings.[936, 942] Coverings should be removed and discarded while the health-care worker is still gloved.[936, 942] The health-care worker, after ungloving and performing hand hygiene, must cover these surfaces with clean materials before the next patient encounter.

b. Cleaning Housekeeping Surfaces

Housekeeping surfaces require regular cleaning and removal of soil and dust. Dry conditions favor the persistence of gram-positive cocci (e.g., coagulase-negative *Staphylococcus* spp.) in dust and on surfaces, whereas moist, soiled environments favor the growth and persistence of gram-negative bacilli.[948, 971, 972] Fungi are also present on dust and proliferate in moist, fibrous material.

Most, if not all, housekeeping surfaces need to be cleaned only with soap and water or a detergent/disinfectant, depending on the nature of the surface and the type and degree of contamination. Cleaning and disinfection schedules and methods vary according to the area of the health-care facility, type of surface to be cleaned, and the amount and type of soil present. Disinfectant/detergent formulations registered by EPA are used for environmental surface cleaning, but the actual physical removal of microorganisms and soil by wiping or scrubbing is probably as important, if not more so, than any antimicrobial effect of the cleaning agent used.[973] Therefore, cost, safety, product-surface compatibility, and acceptability by housekeepers can be the main criteria for selecting a registered agent. If using a proprietary detergent/disinfectant, the manufacturers' instructions for appropriate use

of the product should be followed.[974] Consult the products' material safety data sheets (MSDS) to determine appropriate precautions to prevent hazardous conditions during product application. Personal protective equipment (PPE) used during cleaning and housekeeping procedures should be appropriate to the task.

Housekeeping surfaces can be divided into two groups – those with minimal hand-contact (e.g., floors, and ceilings) and those with frequent hand-contact ("high touch surfaces"). The methods, thoroughness, and frequency of cleaning and the products used are determined by health-care facility policy.[6] However, high-touch housekeeping surfaces in patient-care areas (e.g., doorknobs, bedrails, light switches, wall areas around the toilet in the patient's room, and the edges of privacy curtains) should be cleaned and/or disinfected more frequently than surfaces with minimal hand contact. Infection-control practitioners typically use a risk-assessment approach to identify high-touch surfaces and then coordinate an appropriate cleaning and disinfecting strategy and schedule with the housekeeping staff.

Horizontal surfaces with infrequent hand contact (e.g., window sills and hard-surface flooring) in routine patient-care areas require cleaning on a regular basis, when soiling or spills occur, and when a patient is discharged from the facility.[6] Regular cleaning of surfaces and decontamination, as needed, is also advocated to protect potentially exposed workers.[967] Cleaning of walls, blinds, and window curtains is recommended when they are visibly soiled.[972, 973, 975] Disinfectant fogging is not recommended for general infection control in routine patient-care areas.[2, 976] Further, paraformaldehyde, which was once used in this application, is no longer registered by EPA for this purpose. Use of paraformaldehyde in these circumstances requires either registration or an exemption issued by EPA under the Federal Insecticide, Fungicide, and Rodenticide Act (FIFRA). Infection control, industrial hygienists, and environmental services supervisors should assess the cleaning procedures, chemicals used, and the safety issues to determine if a temporary relocation of the patient is needed when cleaning in the room.

Extraordinary cleaning and decontamination of floors in health-care settings is unwarranted. Studies have demonstrated that disinfection of floors offers no advantage over regular detergent/water cleaning and has minimal or no impact on the occurrence of health-care–associated infections.[947, 948, 977–980] Additionally, newly cleaned floors become rapidly recontaminated from airborne microorganisms and those transferred from shoes, equipment wheels, and body substances.[971, 975, 981] Nevertheless, health-care institutions or contracted cleaning companies may choose to use an EPA-registered detergent/disinfectant for cleaning low-touch surfaces (e.g., floors) in patient-care areas because of the difficulty that personnel may have in determining if a spill contains blood or body fluids (requiring a detergent/disinfectant for clean-up) or when a multi-drug resistant organism is likely to be in the environment. Methods for cleaning non-porous floors include wet mopping and wet vacuuming, dry dusting with electrostatic materials, and spray buffing.[973, 982–984] Methods that produce minimal mists and aerosols or dispersion of dust in patient-care areas are preferred.[9, 20, 109, 272]

Part of the cleaning strategy is to minimize contamination of cleaning solutions and cleaning tools. Bucket solutions become contaminated almost immediately during cleaning, and continued use of the solution transfers increasing numbers of microorganisms to each subsequent surface to be cleaned.[971, 981, 985] Cleaning solutions should be replaced frequently. A variety of "bucket" methods have been devised to address the frequency with which cleaning solutions are replaced.[986, 987] Another source of contamination in the cleaning process is the cleaning cloth or mop head, especially if left soaking in dirty cleaning solutions.[971, 988–990] Laundering of cloths and mop heads after use and allowing them to dry before re-use can help to minimize the degree of contamination.[990] A simplified approach to cleaning involves replacing soiled cloths and mop heads with clean items each time a bucket of detergent/disinfectant is emptied and replaced with fresh, clean solution (B. Stover, Kosair Children's Hospital, 2000). Disposable cleaning cloths and mop heads are an alternative option, if costs permit.

Another reservoir for microorganisms in the cleaning process may be dilute solutions of the detergents or disinfectants, especially if the working solution is prepared in a dirty container, stored for long periods of time, or prepared incorrectly.[547] Gram-negative bacilli (e.g., *Pseudomonas* spp. and *Serratia marcescens*) have been detected in solutions of some disinfectants (e.g., phenolics and quaternary ammonium compounds).[547, 991] Contemporary EPA registration regulations have helped to minimize this problem by asking manufacturers to provide potency data to support label claims for detergent/disinfectant properties under real- use conditions (e.g., diluting the product with tap water instead of distilled water). Application of contaminated cleaning solutions, particularly from small-quantity aerosol spray bottles or with equipment that might generate aerosols during operation, should be avoided, especially in high-risk patient areas.[992, 993] Making sufficient fresh cleaning solution for daily cleaning, discarding any remaining solution, and drying out the container will help to minimize the degree of bacterial contamination. Containers that dispense liquid as opposed to spray-nozzle dispensers (e.g., quart-sized dishwashing liquid bottles) can be used to apply detergent/disinfectants to surfaces and then to cleaning cloths with minimal aerosol generation. A pre-mixed, "ready-to-use" detergent/disinfectant solution may be used if available.

c. Cleaning Special Care Areas

Guidelines have been published regarding cleaning strategies for isolation areas and operating rooms.[6, 7] The basic strategies for areas housing immunosuppressed patients include a) wet dusting horizontal surfaces daily with cleaning cloths pre-moistened with detergent or an EPA-registered hospital disinfectant or disinfectant wipes;[94, 98463] b) using care when wet dusting equipment and surfaces above the patient to avoid patient contact with the detergent/disinfectant; c) avoiding the use of cleaning equipment that produces mists or aerosols; d) equipping vacuums with HEPA filters, especially for the exhaust, when used in any patient-care area housing immunosuppressed patients;[9, 94, 986] and e) regular cleaning and maintenance of equipment to ensure efficient particle removal. When preparing the cleaning cloths for wet-dusting, freshly prepared solutions of detergents or disinfectants should be used rather than cloths that have soaked in such solutions for long periods of time. Dispersal of microorganisms in the air from dust or aerosols is more problematic in these settings than elsewhere in health-care facilities. Vacuum cleaners can serve as dust disseminators if they are not operating properly.[994] Doors to immunosuppressed patients' rooms should be closed when nearby areas are being vacuumed.[9] Bacterial and fungal contamination of filters in cleaning equipment is inevitable, and these filters should be cleaned regularly or replaced as per equipment manufacturer instructions.

Mats with tacky surfaces placed in operating rooms and other patient-care areas only slightly minimize the overall degree of contamination of floors and have little impact on the incidence rate of health-care–associated infection in general.[351, 971, 983] An exception, however, is the use of tacky mats inside the entry ways of cordoned-off construction areas inside the health-care facility; these mats help to minimize the intrusion of dust into patient-care areas.

Special precautions for cleaning incubators, mattresses, and other nursery surfaces have been recommended to address reports of hyperbilirubinemia in newborns linked to inadequately diluted solutions of phenolics and poor ventilation.[995–997] These medical conditions have not, however, been associated with the use of properly prepared solutions of phenolics. Non-porous housekeeping surfaces in neonatal units can be disinfected with properly diluted or pre-mixed phenolics, followed by rinsing with clean water.[997] However, phenolics are not recommended for cleaning infant bassinets and incubators during the stay of the infant. Infants who remain in the nursery for an extended period should be moved periodically to freshly cleaned and disinfected bassinets and incubators.[997] If phenolics are used for cleaning bassinets and incubators after they have been vacated, the surfaces should be rinsed thoroughly with water and dried before either piece of equipment is reused. Cleaning

and disinfecting protocols should allow for the full contact time specified for the product used. Bassinet mattresses should be replaced, however, if the mattress cover surface is broken.[997]

3. Cleaning Strategies for Spills of Blood and Body Substances

Neither HBV, HCV, nor HIV has ever been transmitted from a housekeeping surface (i.e., floors, walls, or countertops). Nonetheless, prompt removal and surface disinfection of an area contaminated by either blood or body substances are sound infection-control practices and OSHA requirements.[967]

Studies have demonstrated that HIV is inactivated rapidly after being exposed to commonly used chemical germicides at concentrations that are much lower than those used in practice.[998–1003] HBV is readily inactivated with a variety of germicides, including quaternary ammonium compounds.[1004] Embalming fluids (e.g., formaldehyde) are also capable of completely inactivating HIV and HBV.[1005, 1006] OSHA has revised its regulation for disinfecting spills of blood or other potentially infectious material to include proprietary products whose label includes inactivation claims for HBV and HIV, provided that such surfaces have not become contaminated with agent(s) or volumes of or concentrations of agent(s) for which a higher level of disinfection is recommended.[1007] These registered products are listed in EPA's List D – *Registered Antimicrobials Effective Against Hepatitis B Virus and Human HIV-1*, which may include products tested against duck hepatitis B virus (DHBV) as a surrogate for HBV.[1008, 1009] Additional lists of interest include EPA's List C –*Registered Antimicrobials Effective Against Human HIV-1* and EPA's List E – *Registered Antimicrobials Effective Against Mycobacterium spp., Hepatitis B Virus, and Human HIV-1*.

Sodium hypochlorite solutions are inexpensive and effective broad-spectrum germicidal solutions.[1010, 1011] Generic sources of sodium hypochlorite include household chlorine bleach or reagent grade chemical. Concentrations of sodium hypochlorite solutions with a range of 5,000–6,150 ppm (1:10 v/v dilution of household bleaches marketed in the United States) to 500–615 ppm (1:100 v/v dilution) free chlorine are effective depending on the amount of organic material (e.g., blood, mucus, and urine) present on the surface to be cleaned and disinfected.[1010, 1011] EPA-registered chemical germicides may be more compatible with certain materials that could be corroded by repeated exposure to sodium hypochlorite, especially the 1:10 dilution. Appropriate personal protective equipment (e.g., gloves and goggles) should be worn when preparing and using hypochlorite solutions or other chemical germicides.[967]

Despite laboratory evidence demonstrating adequate potency against bloodborne pathogens (e.g., HIV and HBV), many chlorine bleach products available in grocery and chemical-supply stores are not registered by the EPA for use as surface disinfectants. Use of these chlorine products as surface disinfectants is considered by the EPA to be an "unregistered use." EPA encourages the use of registered products because the agency reviews them for safety and performance when the product is used according to label instructions. When unregistered products are used for surface disinfection, users do so at their own risk.

Strategies for decontaminating spills of blood and other body fluids differ based on the setting in which they occur and the volume of the spill.[1010] In patient-care areas, workers can manage small spills with cleaning and then disinfecting using an intermediate-level germicide or an EPA-registered germicide from the EPA List D or E.[967, 1007] For spills containing large amounts of blood or other body substances, workers should first remove visible organic matter with absorbent material (e.g., disposable paper towels discarded into leak-proof, properly labeled containment) and then clean and decontaminate the area.[1002, 1003, 1012] If the surface is nonporous and a generic form of a sodium hypochlorite solution is used (e.g., household bleach), a 1:100 dilution is appropriate for decontamination assuming that a) the

worker assigned to clean the spill is wearing gloves and other personal protective equipment appropriate to the task, b) most of the organic matter of the spill has been removed with absorbent material, and c) the surface has been cleaned to remove residual organic matter. A recent study demonstrated that even strong chlorine solutions (i.e., 1:10 dilution of chlorine bleach) may fail to totally inactivate high titers of virus in large quantities of blood, but in the absence of blood these disinfectants can achieve complete viral inactivation.[1011] This evidence supports the need to remove most organic matter from a large spill before final disinfection of the surface. Additionally, EPA-registered proprietary disinfectant label claims are based on use on a pre-cleaned surface.[951, 954]

Managing spills of blood, body fluids, or other infectious materials in clinical, public health, and research laboratories requires more stringent measures because of a) the higher potential risk of disease transmission associated with large volumes of blood and body fluids and b) high numbers of microorganisms associated with diagnostic cultures. The use of an intermediate-level germicide for routine decontamination in the laboratory is prudent.[954] Recommended practices for managing large spills of concentrated infectious agents in the laboratory include a) confining the contaminated area, b) flooding the area with a liquid chemical germicide before cleaning, and c) decontaminating with fresh germicidal chemical of at least intermediate-level disinfectant potency.[1010] A suggested technique when flooding the spill with germicide is to lay absorbent material down on the spill and apply sufficient germicide to thoroughly wet both the spill and the absorbent material.[1013] If using a solution of household chlorine bleach, a 1:10 dilution is recommended for this purpose. EPA-registered germicides should be used according to the manufacturers' instructions for use dilution and contact time. Gloves should be worn during the cleaning and decontamination procedures in both clinical and laboratory settings. PPE in such a situation may include the use of respiratory protection (e.g., an N95 respirator) if clean-up procedures are expected to generate infectious aerosols. Protocols for cleaning spills should be developed and made available on record as part of good laboratory practice.[1013] Workers in laboratories and in patient-care areas of the facility should receive periodic training in environmental-surface infection-control strategies and procedures as part of an overall infection-control and safety curriculum.

4. Carpeting and Cloth Furnishings

a. Carpeting

Carpeting has been used for more than 30 years in both public and patient-care areas of health-care facilities. Advantages of carpeting in patient-care areas include a) its noise-limiting characteristics; b) the "humanizing" effect on health care; and c) its contribution to reductions in falls and resultant injuries, particularly for the elderly.[1014–1016] Compared to hard-surface flooring, however, carpeting is harder to keep clean, especially after spills of blood and body substances. It is also harder to push equipment with wheels (e.g., wheelchairs, carts, and gurneys) on carpeting.

Several studies have documented the presence of diverse microbial populations, primarily bacteria and fungi, in carpeting;[111, 1017–1024] the variety and number of microorganisms tend to stabilize over time. New carpeting quickly becomes colonized, with bacterial growth plateauing after about 4 weeks.[1019] Vacuuming and cleaning the carpeting can temporarily reduce the numbers of bacteria, but these populations soon rebound and return to pre-cleaning levels.[1019, 1020, 1023] Bacterial contamination tends to increase with higher levels of activity.[1018–1020, 1025] Soiled carpeting that is or remains damp or wet provides an ideal setting for the proliferation and persistence of gram-negative bacteria and fungi.[1026] Carpeting that remains damp should be removed, ideally within 72 hours.

Despite the evidence of bacterial growth and persistence in carpeting, only limited epidemiologic evidence demonstrates that carpets influence health-care–associated infection rates in areas housing

immunocompetent patients.[1023, 1025, 1027] This guideline, therefore, includes no recommendations against the use of carpeting in these areas. Nonetheless, avoiding the use of carpeting is prudent in areas where spills are likely to occur (e.g., laboratories, areas around sinks, and janitor closets) and where patients may be at greater risk of infection from airborne environmental pathogens (e.g., HSCT units, burn units, ICUs, and ORs).[111, 1028] An outbreak of aspergillosis in an HSCT unit was recently attributed to carpet contamination and a particular method of carpet cleaning.[111] A window in the unit had been opened repeatedly during the time of a nearby building fire, which allowed fungal spore intrusion into the unit. After the window was sealed, the carpeting was cleaned using a "bonnet buffing" machine, which dispersed *Aspergillus* spores into the air.[111] Wet vacuuming was instituted, replacing the dry cleaning method used previously; no additional cases of invasive aspergillosis were identified.

The care setting and the method of carpet cleaning are important factors to consider when attempting to minimize or prevent production of aerosols and dispersal of carpet microorganisms into the air.[94, 111] Both vacuuming and shampooing or wet cleaning with equipment can disperse microorganisms to the air.[111, 994] Vacuum cleaners should be maintained to minimize dust dispersal in general, and be equipped with HEPA filters, especially for use in high-risk patient-care areas.[9, 94, 986] Some formulations of carpet-cleaning chemicals, if applied or used improperly, can be dispersed into the air as a fine dust capable of causing respiratory irritation in patients and staff.[1029] Cleaning equipment, especially those that engage in wet cleaning and extraction, can become contaminated with waterborne organisms (e.g., *Pseudomonas aeruginosa*) and serve as a reservoir for these organisms if this equipment is not properly maintained. Substantial numbers of bacteria can then be transferred to carpeting during the cleaning process.[1030] Therefore, keeping the carpet cleaning equipment in good repair and allowing such equipment to dry between uses is prudent.

Carpet cleaning should be performed on a regular basis determined by internal policy. Although spills of blood and body substances on non-porous surfaces require prompt spot cleaning using standard cleaning procedures and application of chemical germicides,[967] similar decontamination approaches to blood and body substance spills on carpeting can be problematic from a regulatory perspective.[1031] Most, if not all, modern carpet brands suitable for public facilities can tolerate the activity of a variety of liquid chemical germicides. However, according to OSHA, carpeting contaminated with blood or other potentially infectious materials can not be fully decontaminated.[1032] Therefore, facilities electing to use carpeting for high-activity patient-care areas may choose carpet tiles in areas at high risk for spills.[967, 1032] In the event of contamination with blood or other body substances, carpet tiles can be removed, discarded, and replaced. OSHA also acknowledges that only minimal direct skin contact occurs with carpeting, and therefore, employers are expected to make reasonable efforts to clean and sanitize carpeting using carpet detergent/cleaner products.[1032]

Over the last few years, some carpet manufacturers have treated their products with fungicidal and/or bactericidal chemicals. Although these chemicals may help to reduce the overall numbers of bacteria or fungi present in carpet, their use does not preclude the routine care and maintenance of the carpeting. Limited evidence suggests that chemically treated carpet may have helped to keep health-care–associated aspergillosis rates low in one HSCT unit,[111] but overall, treated carpeting has not been shown to prevent the incidence of health-care–associated infections in care areas for immunocompetent patients.

b. Cloth Furnishings

Upholstered furniture and furnishings are becoming increasingly common in patient-care areas. These furnishings range from simple cloth chairs in patients' rooms to a complete decorating scheme that gives the interior of the facility more the look of an elegant hotel.[1033] Even though pathogenic microorganisms have been isolated from the surfaces of cloth chairs, no epidemiologic evidence suggests that general patient-care areas with cloth furniture pose increased risks of health-care–

associated infection compared with areas that contain hard-surfaced furniture.[1034, 1035] Allergens (e.g., dog and cat dander) have been detected in or on cloth furniture in clinics and elsewhere in hospitals in concentrations higher than those found on bed linens.[1034, 1035] These allergens presumably are transferred from the clothing of visitors. Researchers have therefore suggested that cloth chairs should be vacuumed regularly to keep the dust and allergen levels to a minimum. This recommendation, however, has generated concerns that aerosols created from vacuuming could place immunocompromised patients or patients with preexisting lung disease (e.g., asthma) at risk for development of health-care–associated, environmental airborne disease.[9, 20, 109, 988] Recovering worn, upholstered furniture (especially the seat cushion) with covers that are easily cleaned (e.g., vinyl), or replacing the item is prudent; minimizing the use of upholstered furniture and furnishings in any patient-care areas where immunosuppressed patients are located (e.g., HSCT units) reduces the likelihood of disease.[9]

5. Flowers and Plants in Patient-Care Areas

Fresh flowers, dried flowers, and potted plants are common items in health-care facilities. In 1974, clinicians isolated an *Erwinia* sp. post mortem from a neonate diagnosed with fulminant septicemia, meningitis, and respiratory distress syndrome.[1038] Because *Erwinia* spp. are plant pathogens, plants brought into the delivery room were suspected to be the source of the bacteria, although the case report did not definitively establish a direct link. Several subsequent studies evaluated the numbers and diversity of microorganisms in the vase water of cut flowers. These studies revealed that high concentrations of bacteria, ranging from 10^4–10^{10} CFU/mL, were often present, especially if the water was changed infrequently.[515, 702, 1039] The major group of microorganisms in flower vase water was gram-negative bacteria, with *Pseudomonas aeruginosa* the most frequently isolated organism.[515, 702, 1039, 1040] *P. aeruginosa* was also the primary organism directly isolated from chrysanthemums and other potted plants.[1041, 1042] However, flowers in hospitals were not significantly more contaminated with bacteria compared with flowers in restaurants or in the home.[702] Additionally, no differences in the diversity and degree of antibiotic resistance of bacteria have been observed in samples isolated from hospital flowers versus those obtained from flowers elsewhere.[702]

Despite the diversity and large numbers of bacteria associated with flower-vase water and potted plants, minimal or no evidence indicates that the presence of plants in immunocompetent patient-care areas poses an increased risk of health-care–associated infection.[515] In one study involving a limited number of surgical patients, no correlation was observed between bacterial isolates from flowers in the area and the incidence and etiology of postoperative infections among the patients.[1040] Similar conclusions were reached in a study that examined the bacteria found in potted plants.[1042] Nonetheless, some precautions for general patient-care settings should be implemented, including a) limiting flower and plant care to staff with no direct patient contact, b) advising health-care staff to wear gloves when handling plants, c) washing hands after handling plants, d) changing vase water every 2 days and discharging the water into a sink outside the immediate patient environment, and e) cleaning and disinfecting vases after use.[702]

Some researchers have examined the possibility of adding a chemical germicide to vase water to control bacterial populations. Certain chemicals (e.g., hydrogen peroxide and chlorhexidine) are well tolerated by plants.[1040, 1043, 1044] Use of these chemicals, however, was not evaluated in studies to assess impact on health-care–associated infection rates. Modern florists now have a variety of products available to add to vase water to extend the life of cut flowers and to minimize bacterial clouding of the water.

Flowers (fresh and dried) and ornamental plants, however, may serve as a reservoir of *Aspergillus* spp., and dispersal of conidiospores into the air from this source can occur.[109] Health-care–associated outbreaks of invasive aspergillosis reinforce the importance of maintaining an environment as free of

Aspergillus spp. spores as possible for patients with severe, prolonged neutropenia. Potted plants, fresh-cut flowers, and dried flower arrangements may provide a reservoir for these fungi as well as other fungal species (e.g., *Fusarium* spp.).[109, 1045, 1046] Researchers in one study of bacteria and flowers suggested that flowers and vase water should be avoided in areas providing care to medically at-risk patients (e.g., oncology patients and transplant patients), although this study did not attempt to correlate the observations of bacterial populations in the vase water with the incidence of health-care–associated infections.[515] Another study using molecular epidemiology techniques demonstrated identical *Aspergillus terreus* types among environmental and clinical specimens isolated from infected patients with hematological malignancies.[1046] Therefore, attempts should be made to exclude flowers and plants from areas where immunosuppressed patients are be located (e.g., HSCT units).[9, 1046]

6. Pest Control

Cockroaches, flies and maggots, ants, mosquitoes, spiders, mites, midges, and mice are among the typical arthropod and vertebrate pest populations found in health-care facilities. Insects can serve as agents for the mechanical transmission of microorganisms, or as active participants in the disease transmission process by serving as a vector.[1047–1049] Arthropods recovered from health-care facilities have been shown to carry a wide variety of pathogenic microorganisms.[1050–1056] Studies have suggested that the diversity of microorganisms associated with insects reflects the microbial populations present in the indoor health-care environment; some pathogens encountered in insects from hospitals were either absent from or present to a lesser degree in insects trapped from residential settings.[1057–1060] Some of the microbial populations associated with insects in hospitals have demonstrated resistance to antibiotics.[1048, 1059, 1061–1063]

Insect habitats are characterized by warmth, moisture, and availability of food.[1064] Insects forage in and feed on substrates, including but not limited to food scraps from kitchens/cafeteria, foods in vending machines, discharges on dressings either in use or discarded, other forms of human detritis, medical wastes, human wastes, and routine solid waste.[1057–1061] Cockroaches, in particular, have been known to feed on fixed sputum smears in laboratories.[1065, 1066] Both cockroaches and ants are frequently found in the laundry, central sterile supply departments, and anywhere in the facility where water or moisture is present (e.g., sink traps, drains and janitor closets). Ants will often find their way into sterile packs of items as they forage in a warm, moist environment.[1057] Cockroaches and other insects frequent loading docks and other areas with direct access to the outdoors.

Although insects carry a wide variety of pathogenic microorganisms on their surfaces and in their gut, the direct association of insects with disease transmission (apart from vector transmission) is limited, especially in health-care settings; the presence of insects in itself likely does not contribute substantially to health-care–associated disease transmission in developed countries. However, outbreaks of infection attributed to microorganisms carried by insects may occur because of infestation coupled with breaks in standard infection-control practices.[1063] Studies have been conducted to examine the role of houseflies as possible vectors for shigellosis and other forms of diarrheal disease in non-health–care settings.[1046, 1067] When control measures aimed at reducing the fly population density were implemented, a concomitant reduction in the incidence of diarrheal infections, carriage of *Shigella* organisms, and mortality caused by diarrhea among infants and young children was observed.

Myiasis is defined as a parasitosis in which the larvae of any of a variety of flies use living or necrotic tissue or body substances of the host as a nutritional source.[1068] Larvae from health-care–acquired myiasis have been observed in nares, wounds, eyes, ears, sinuses, and the external urogenital structures.[1069–1071] Patients with this rare condition are typically older adults with underlying medical conditions (e.g., diabetes, chronic wounds, and alcoholism) who have a decreased capacity to ward off

the flies. Persons with underlying conditions who live or travel to tropical regions of the world are especially at risk.[1070, 1071] Cases occur in the summer and early fall months in temperate climates when flies are most active.[1071] An environmental assessment and review of the patient's history are necessary to verify that the source of the myiasis is health-care–acquired and to identify corrective measures.[1069, 1072] Simple prevention measures (e.g., installing screens on windows) are important in reducing the incidence of myiasis.[1072]

From a public health and hygiene perspective, arthropod and vertebrate pests should be eradicated from all indoor environments, including health-care facilities.[1073, 1074] Modern approaches to institutional pest management usually focus on a) eliminating food sources, indoor habitats, and other conditions that attract pests; b) excluding pests from the indoor environments; and c) applying pesticides as needed.[1075] Sealing windows in modern health-care facilities helps to minimize insect intrusion. When windows need to be opened for ventilation, ensuring that screens are in good repair and closing doors to the outside can help with pest control. Insects should be kept out of all areas of the health-care facility, especially ORs and any area where immunosuppressed patients are located. A pest-control specialist with appropriate credentials can provide a regular insect-control program that is tailored to the needs of the facility and uses approved chemicals and/or physical methods. Industrial hygienists can provide information on possible adverse reactions of patients and staff to pesticides and suggest alternative methods for pest control, as needed.

7. Special Pathogen Concerns

a. Antibiotic-Resistant Gram-Positive Cocci

Vancomycin-resistant enterococci (VRE), methicillin-resistant *Staphylococcus aureus* (MRSA), and *S. aureus* with intermediate levels of resistance to glycopeptide antibiotics (vancomycin intermediate resistant *S. aureus* [VISA] or glycopeptide intermediate resistant *S. aureus* [GISA]) represent crucial and growing concerns for infection control. Although the term GISA is technically a more accurate description of the strains isolated to date (most of which are classified as having intermediate resistance to both vancomycin and teicoplanin), the term "glycopeptide" may not be recognized by many clinicians. Thus, the label of VISA, which emphasizes a change in minimum inhibitory concentration (MICs) to vancomycin, is similar to that of VRE and is more meaningful to clinicians.[1076] According to National Nosocomial Infection Surveillance (NNIS) statistics for infections acquired among ICU patients in the United States in 1999, 52.3% of infections resulting from *S. aureus* were identified as MRSA infections, and 25.2% of enterococcal infections were attributed to VRE. These figures reflect a 37% and a 43% increase, respectively, since 1994–1998.[1077]

People represent the primary reservoir of *S. aureus*.[1078] Although *S. aureus* has been isolated from a variety of environmental surfaces (e.g., stethoscopes, floors, charts, furniture, dry mops, and hydrotherapy tanks), the role of environmental contamination in transmission of this organism in health care appears to be minimal.[1079–1082] *S. aureus* contamination of surfaces and tanks within burn therapy units, however, may be a major factor in the transmission of infection among burn patients.[1083]

Colonized patients are the principal reservoir of VRE, and patients who are immunosuppressed (e.g., transplant patients) or otherwise medically at-risk (e.g., ICU patients, cardio-thoracic surgical patients, patients previously hospitalized for extended periods, and those having received multi-antimicrobial or vancomycin therapy) are at greatest risk for VRE colonization.[1084–1087] The mechanisms by which cross-colonization take place are not well defined, although recent studies have indicated that both MRSA and VRE may be transmitted either a) directly from patient to patient, b) indirectly by transient carriage on the hands of health-care workers,[1088–1091] or c) by hand transfer of these gram-positive organisms from contaminated environmental surfaces and patient-care equipment.[1084, 1087, 1092–1097] In

one survey, hand carriage of VRE in workers in a long-term care facility ranged from 13%–41%.[1098] Many of the environmental surfaces found to be contaminated with VRE in outbreak investigations have been those that are touched frequently by the patient or the health-care worker.[1099] Such high-touch surfaces include bedrails, doorknobs, bed linens, gowns, overbed tables, blood pressure cuffs, computer table, bedside tables, and various medical equipment.[22, 1087, 1094, 1095, 1100–1102] Contamination of environmental surfaces with VRE generally occurs in clinical laboratories and areas where colonized patients are present,[1087, 1092, 1094, 1095, 1103] but the potential for contamination increases when such patients have diarrhea[1087] or have multiple body-site colonization.[1104] Additional factors that can be important in the dispersion of these pathogens to environmental surfaces are misuse of glove techniques by health-care workers (especially when cleaning fecal contamination from surfaces) and patient, family, and visitor hygiene.

Interest in the importance of environmental reservoirs of VRE increased when laboratory studies demonstrated that enterococci can persist in a viable state on dry environmental surfaces for extended periods of time (7 days to 4 months)[1099, 1105] and multiple strains can be identified during extensive periods of surveillance.[1104] VRE can be recovered from inoculated hands of health-care workers (with or without gloves) for up to 60 minutes.[22] The presence of either MRSA, VISA, or VRE on environmental surfaces, however, does not mean that patients in the contaminated areas will become colonized. Strict adherence to hand hygiene/handwashing and the proper use of barrier precautions help to minimize the potential for spread of these pathogens. Published recommendations for preventing the spread of vancomycin resistance address isolation measures, including patient cohorting and management of patient-care items.[5] Direct patient-care items (e.g., blood pressure cuffs) should be disposable whenever possible when used in contact isolation settings for patients with multiply resistant microorganisms.[1102]

Careful cleaning of patient rooms and medical equipment contributes substantially to the overall control of MRSA, VISA, or VRE transmission. The major focus of a control program for either VRE or MRSA should be the prevention of hand transfer of these organisms. Routine cleaning and disinfection of the housekeeping surfaces (e.g., floors and walls) and patient-care surfaces (e.g., bedrails) should be adequate for inactivation of these organisms. Both MRSA and VRE are susceptible to several EPA-registered low- and intermediate-level disinfectants (e.g., alcohols, sodium hypochlorite, quaternary ammonium compounds, phenolics, and iodophors) at recommended use dilutions for environmental surface disinfection.[1103, 1106–1109] Additionally, both VRE and vancomycin-sensitive enterococci are equally sensitive to inactivation by chemical germicides,[1106, 1107, 1109] and similar observations have been made when comparing the germicidal resistance of MRSA to that of either methicillin-sensitive *S. aureus* (MSSA) or VISA.[1110] The use of stronger solutions of disinfectants for inactivation of either VRE, MRSA, or VISA is not recommended based on the organisms' resistance to antibiotics.[1110–1112] VRE from clinical specimens have exhibited some measure of increased tolerance to heat inactivation in temperature ranges <212ºF (<100ºC);[1106, 1113] however, the clinical significance of these observations is unclear because the role of cleaning the surface or item prior to heat treatment was not evaluated. Although routine environmental sampling is not recommended, laboratory surveillance of environmental surfaces during episodes when VRE contamination is suspected can help determine the effectiveness of the cleaning and disinfecting procedures. Environmental culturing should be approved and supervised by the infection-control program in collaboration with the clinical laboratory.[1084, 1087, 1088, 1092, 1096]

Two cases of wound infections associated with vancomycin-resistant *Staphylococcus aureus* (VRSA) determined to be resistant by NCCLS standards for sensitivity/resistance testing were identified in Michigan and Pennsylvania in 2002.[1114, 1115] These represented isolated cases, and neither the family members nor the health-care providers of these case-patients had evidence of colonization or infection with VRSA. Conventional environmental infection-control measures (i.e., cleaning and then

disinfecting surfaces using EPA-registered disinfectants with label claims for *S. aureus*) were used during the environmental investigation of these two cases;[1110–1112] however, studies have yet to evaluate the potential intrinsic resistance of these VRSA strains to surface disinfectants.

Standard procedures during terminal cleaning and disinfection of surfaces, if performed incorrectly, may be inadequate for the elimination of VRE from patient rooms.[1113, 1116–1118] Given the sensitivity of VRE to hospital disinfectants, current disinfecting protocols should be effective if they are diligently carried out and properly performed. Health-care facilities should be sure that housekeeping staff use correct procedures for cleaning and disinfecting surfaces in VRE-contaminated areas, which include using sufficient amounts of germicide at proper use dilution and allowing adequate contact time.[1118]

b. Clostridium difficile

Clostridium difficile is the most frequent etiologic agent for health-care–associated diarrhea.[1119, 1120] In one hospital, 30% of adults who developed health-care–associated diarrhea were positive for *C. difficile*.[1121] One recent study employing PCR-ribotyping techniques demonstrated that cases of *C. difiicile*-acquired diarrhea occurring in the hospital included patients whose infections were attributed to endogenous *C. difficile* strains and patients whose illnesses were considered to be health-care–associated infections.[1122] Most patients remain asymptomatic after infection, but the organism continues to be shed in their stools. Risk factors for acquiring *C. difficile*-associated infection include a) exposure to antibiotic therapy, particularly with beta-lactam agents;[1123] b) gastrointestinal procedures and surgery;[1124] c) advanced age; and d) indiscriminate use of antibiotics.[1125–1128] Of all the measures that have been used to prevent the spread of *C. difficile*-associated diarrhea, the most successful has been the restriction of the use of antimicrobial agents.[1129, 1130]

C. difficile is an anaerobic, gram-positive bacterium. Normally fastidious in its vegetative state, it is capable of sporulating when environmental conditions no longer support its continued growth. The capacity to form spores enables the organism to persist in the environment (e.g., in soil and on dry surfaces) for extended periods of time. Environmental contamination by this microorganism is well known, especially in places where fecal contamination may occur.[1131] The environment (especially housekeeping surfaces) rarely serves as a direct source of infection for patients.[1024, 1132–1136] However, direct exposure to contaminated patient-care items (e.g., rectal thermometers) and high-touch surfaces in patients' bathrooms (e.g., light switches) have been implicated as sources of infection.[1130, 1135, 1136, 1138]

Transfer of the pathogen to the patient via the hands of health-care workers is thought to be the most likely mechanism of exposure.[24, 1133, 1139] Standard isolation techniques intended to minimize enteric contamination of patients, health-care–workers' hands, patient-care items, and environmental surfaces have been published.[1140] Handwashing remains the most effective means of reducing hand contamination. Proper use of gloves is an ancillary measure that helps to further minimize transfer of these pathogens from one surface to another.

The degree to which the environment becomes contaminated with *C. difficile* spores is proportional to the number of patients with *C. difficile*-associated diarrhea,[24, 1132, 1135] although asymptomatic, colonized patients may also serve as a source of contamination. Few studies have examined the use of specific chemical germicides for the inactivation of *C. difficile* spores, and no well-controlled trials have been conducted to determine efficacy of surface disinfection and its impact on health-care–associated diarrhea. Some investigators have evaluated the use of chlorine-containing chemicals (e.g., 1,000 ppm hypochlorite at recommended use-dilution, 5,000 ppm sodium hypochlorite [1:10 v/v dilution], 1:100 v/v dilutions of unbuffered hypochlorite, and phosphate-buffered hypochlorite [1,600 ppm]). One of the studies demonstrated that the number of contaminated environmental sites was reduced by half,[1135] whereas another two studies demonstrated declines in health-care–associated *C. difficile* infections in a HSCT unit[1141] and in two geriatric medical units[1142] during a period of hypochlorite use. The presence

of confounding factors, however, was acknowledged in one of these studies.[1142] The recommended approach to environmental infection control with respect to *C. difficile* is meticulous cleaning followed by disinfection using hypochlorite-based germicides as appropriate.[952, 1130, 1143] However, because no EPA-registered surface disinfectants with label claims for inactivation of *C. difficile* spores are available, the recommendation is based on the best available evidence from the scientific literature.

c. Respiratory and Enteric Viruses in Pediatric-Care Settings

Although the viruses mentioned in this guideline are not unique to the pediatric-care setting in health-care facilities, their prevalence in these areas, especially during the winter months, is substantial. Children (particularly neonates) are more likely to develop infection and substantial clinical disease from these agents compared with adults and therefore are more likely to require supportive care during their illness.

Common respiratory viruses in pediatric-care areas include rhinoviruses, respiratory syncytial virus (RSV), adenoviruses, influenza viruses, and parainfluenza viruses. Transmission of these viruses occurs primarily via direct contact with small-particle aerosols or via hand contamination with respiratory secretions that are then transferred to the nose or eyes. Because transmission primarily requires close personal contact, contact precautions are appropriate to interrupt transmission.[6] Hand contamination can occur from direct contact with secretions or indirectly from touching high-touch environmental surfaces that have become contaminated with virus from large droplets. The indirect transfer of virus from one persion to other via hand contact with frequently-touched fomites was demonstrated in a study using a bacteriophage whose environmental stability approximated that of human viral pathogens (e.g., poliovirus and parvovirus).[1144] The impact of this mode of transmission with respect to human respiratory- and enteric viruses is dependent on the ability of these agents to survive on environmental surfaces. Infectious RSV has been recovered from skin, porous surfaces, and non-porous surfaces after 30 minutes, 1 hour, and 7 hours, respectively.[1145] Parainfluenza viruses are known to persist for up to 4 hours on porous surfaces and up to 10 hours on non-porous surfaces.[1146] Rhinoviruses can persist on porous surfaces and non-porous surfaces for approximately 1 and 3 hours respectively; study participants in a controlled environment became infected with rhinoviruses after first touching a surface with dried secretions and then touching their nasal or conjunctival mucosa.[1147] Although the efficiency of direct transmission of these viruses from surfaces in uncontrolled settings remains to be defined, these data underscore the basis for maintaining regular protocols for cleaning and disinfecting of high-touch surfaces.

The clinically important enteric viruses encountered in pediatric care settings include enteric adenovirus, astroviruses, caliciviruses, and rotavirus. Group A rotavirus is the most common cause of infectious diarrhea in infants and children. Transmission of this virus is primarily fecal-oral, however, the role of fecally contaminated surfaces and fomites in rotavirus transmission is unclear. During one epidemiologic investigation of enteric disease among children attending day care, rotavirus contamination was detected on 19% of inanimate objects in the center.[1148, 1149] In an outbreak in a pediatric unit, secondary cases of rotavirus infection clustered in areas where children with rotaviral diarrhea were located.[1150] Astroviruses cause gastroenteritis and diarrhea in newborns and young children and can persist on fecally contaminated surfaces for several months during periods of relatively low humidity.[1151, 1152] Outbreaks of small round-structured viruses (i.e., caliciviruses [Norwalk virus and Norwalk-like viruses]) can affect both patients and staff, with attack rates of $\geq$50%.[1153] Routes of person-to-person transmission include fecal-oral spread and aerosols generated from vomiting.[1154–1156] Fecal contamination of surfaces in care settings can spread large amounts of virus to the environment. Studies that have attempted to use low- and intermediate-level disinfectants to inactivate rotavirus suspended in feces have demonstrated a protective effect of high concentrations of organic matter.[1157, 1158] Intermediate-level disinfectants (e.g., alcoholic quaternary ammonium compounds, and chlorine solutions) can be effective in inactivating enteric viruses provided that a cleaning step to remove most of

the organic matter precedes terminal disinfection.[1158] These findings underscore the need for proper cleaning and disinfecting procedures where contamination of environmental surfaces with body substances is likely. EPA-registered surface disinfectants with label claims for these viral agents should be used in these settings. Using disposable, protective barrier coverings may help to minimize the degree of surface contamination.[936]

d. Severe Acute Respiratory Syndrome (SARS) Virus

In November 2002 an atypical pneumonia of unknown etiology emerged in Asia and subsequently developed into an international outbreak of respiratory illness among persons in 29 countries during the first six months of 2003. "Severe acute respiratory syndrome" (SARS) is a viral upper respiratory infection associated with a newly described coronavirus (SARS-associated Co-V [SARS-CoV]). SARS-CoV is an enveloped RNA virus. It is present in high titers in respiratory secretions, stool, and blood of infected persons. The modes of transmission determined from epidemiologic investigations were primarily forms of direct contact (i.e., large droplet aerosolization and person-to-person contact). Respiratory secretions were presumed to be the major source of virus in these situations; airborne transmission of virus has not been completely ruled out. Little is known about the impact of fecal-oral transmission and SARS.

The epidemiology of SARS-CoV infection is not completely understood, and therefore recommended infection control and prevention measures to contain the spread of SARS will evolve as new information becomes available.[1159] At present there is no indication that established strategies for cleaning (i.e., to remove the majority of bioburden) and disinfecting equipment and environmental surfaces need to be changed for the environmental infection control of SARS. In-patient rooms housing SARS patients should be cleaned and disinfected at least daily and at the time of patient transfer or discharge. More frequent cleaning and disinfection may be indicated for high-touch surfaces and following aerosol-producing procedures (e.g., intubation, bronchoscopy, and sputum production). While there are presently no disinfectant products registered by EPA specifically for inactivation of SARS-CoV, EPA-registered hospital disinfectants that are equivalent to low- and intermediate-level germicides may be used on pre-cleaned, hard, non-porous surfaces in accordance with manufacturer's instructions for environmental surface disinfection. Monitoring adherence to guidelines established for cleaning and disinfection is an important component of environmental infection control to contain the spread of SARS.

e. Creutzfeldt-Jakob Disease (CJD) in Patient-Care Areas

Creutzfeldt-Jakob disease (CJD) is a rare, invariably fatal, transmissible spongiform encephalopathy (TSE) that occurs worldwide with an average annual incidence of 1 case per million population.[1160–1162] CJD is one of several TSEs affecting humans; other diseases in this group include kuru, fatal familial insomnia, and Gerstmann-Sträussler-Scheinker syndrome. A TSE that affects a younger population (compared to the age range of CJD cases) has been described primarily in the United Kingdom since 1996.[1163] This variant form of CJD (vCJD) is clinically and neuropathologically distinguishable from classic CJD; epidemiologic and laboratory evidence suggests a causal association for bovine spongiform encephalopathy (BSE [Mad Cow disease]) and vCJD.[1163–1166]

The agent associated with CJD is a prion, which is an abnormal isoform of a normal protein constituent of the central nervous system.[1167–1169] The mechanism by which the normal form of the protein is converted to the abnormal, disease-causing prion is unknown. The tertiary conformation of the abnormal prion protein appears to confer a heightened degree of resistance to conventional methods of sterilization and disinfection.[1170, 1171]

Although about 90% of CJD cases occur sporadically, a limited number of cases are the result of a direct exposure to prion-containing material (usually central nervous system tissue or pituitary

hormones) acquired as a result of health care (iatrogenic cases). These cases have been linked to a) pituitary hormone therapy [from human sources as opposed to hormones prepared through the use of recombinant technology],[1170–1174] b) transplants of either dura mater or corneas,[1175–1181] and c) neurosurgical instruments and depth electrodes.[1182–1185] In the cases involving instruments and depth electrodes, conventional cleaning and terminal reprocessing methods of the day failed to fully inactivate the contaminating prions and are considered inadequate by today's standards.

Prion inactivation studies involving whole tissues and tissue homogenates have been conducted to determine the parameters of physical and chemical methods of sterilization or disinfection necessary for complete inactivation;[1170, 1186–1191] however, the application of these findings to environmental infection control in health-care settings is problematic. No studies have evaluated the effectiveness of medical instrument reprocessing in inactivating prions. Despite a consensus that abnormal prions display some extreme measure of resistance to inactivation by either physical or chemical methods, scientists disagree about the exact conditions needed for sterilization. Inactivation studies utilizing whole tissues present extraordinary challenges to any sterilizing method.[1192] Additionally, the experimental designs of these studies preclude the evaluation of surface cleaning as a part of the total approach to pathogen inactivation.[951, 1192]

Some researchers have recommended the use of either a 1:2 v/v dilution of sodium hypochorite (approximately 20,000 ppm), full-strength sodium hypochlorite (50,000–60,000 ppm), or 1–2 N sodium hydroxide (NaOH) for the inactivation of prions on certain surfaces (e.g., those found in the pathology laboratory).[1170, 1188] Although these chemicals may be appropriate for the decontamination of laboratory, operating-room, or autopsy-room surfaces that come into contact with central nervous system tissue from a known or suspected patient, this approach is not indicated for routine or terminal cleaning of a room previously occupied by a CJD patient. Both chemicals pose hazards for the health-care worker doing the decontamination. NaOH is caustic and should not make contact with the skin. Sodium hypochlorite solutions (i.e., chlorine bleach) can corrode metals (e.g., aluminum). MSDS information should be consulted when attempting to work with concentrated solutions of either chemical. Currently, no EPA-registered products have label claims for prion inactivation; therefore, this guidance is based on the best available evidence from the scientific literature.

Environmental infection-control strategies must based on the principles of the "chain of infection," regardless of the disease of concern.[13] Although CJD is transmissible, it is not highly contagious. All iatrogenic cases of CJD have been linked to a direct exposure to prion-contaminated central nervous system tissue or pituitary hormones. The six documented iatrogenic cases associated with instruments and devices involved neurosurgical instruments and devices that introduced residual contamination directly to the recipient's brain. No evidence suggests that vCJD has been transmitted iatrogenically or that either CJD or vCJD has been transmitted from environmental surfaces (e.g., housekeeping surfaces). Therefore, routine procedures are adequate for terminal cleaning and disinfection of a CJD patient's room. Additionally, in epidemiologic studies involving highly transfused patients, blood was not identified as a source for prion transmission.[1193–1198] Routine procedures for containing, decontaminating, and disinfecting surfaces with blood spills should be adequate for proper infection control in these situations.[951, 1199]

Guidance for environmental infection control in ORs and autopsy areas has been published.[1197, 1199] Hospitals should develop risk-assessment procedures to identify patients with known or suspected CJD in efforts to implement prion-specific infection-control measures for the OR and for instrument reprocessing.[1200] This assessment also should be conducted for older patients undergoing non-lesionous neurosurgery when such procedures are being done for diagnosis. Disposable, impermeable coverings should be used during these autopsies and neurosurgeries to minimize surface contamination. Surfaces that have become contaminated with central nervous system tissue or cerebral spinal fluid should be

cleaned and decontaminated by a) removing most of the tissue or body substance with absorbent materials, b) wetting the surface with a sodium hypochlorite solution containing ≥5,000 ppm or a 1 N NaOH solution, and c) rinsing thoroughly.[951, 1197–1199, 1201] The optimum duration of contact exposure in these instances is unclear. Some researchers recommend a 1-hour contact time on the basis of tissue-inactivation studies,[1197, 1198, 1201] whereas other reviewers of the subject draw no conclusions from this research.[1199] Factors to consider before cleaning a potentially contaminated surface are a) the degree to which gross tissue/body substance contamination can be effectively removed and b) the ease with which the surface can be cleaned.

F. Environmental Sampling

This portion of Part I addresses the basic principles and methods of sampling environmental surfaces and other environmental sources for microorganisms. The applied strategies of sampling with respect to environmental infection control have been discussed in the appropriate preceding subsections.

1. General Principles: Microbiologic Sampling of the Environment

Before 1970, U.S. hospitals conducted regularly scheduled culturing of the air and environmental surfaces (e.g., floors, walls, and table tops).[1202] By 1970, CDC and the American Hospital Association (AHA) were advocating the discontinuation of routine environmental culturing because rates of health-care–associated infection had not been associated with levels of general microbial contamination of air or environmental surfaces, and because meaningful standards for permissible levels of microbial contamination of environmental surfaces or air did not exist.[1203–1205] During 1970–1975, 25% of U.S. hospitals reduced the extent of such routine environmental culturing — a trend that has continued.[1206, 1207]

Random, undirected sampling (referred to as "routine" in previous guidelines) differs from the current practice of targeted sampling for defined purposes.[2, 1204] Previous recommendations against routine sampling were not intended to discourage the use of sampling in which sample collection, culture, and interpretation are conducted in accordance with defined protocols.[2] In this guideline, targeted microbiologic sampling connotes a monitoring process that includes a) a written, defined, multidisciplinary protocol for sample collection and culturing; b) analysis and interpretation of results using scientifically determined or anticipatory baseline values for comparison; and c) expected actions based on the results obtained. Infection control, in conjunction with laboratorians, should assess the health-care facility's capability to conduct sampling and determine when expert consultation and/or services are needed.

Microbiologic sampling of air, water, and inanimate surfaces (i.e., environmental sampling) is an expensive and time-consuming process that is complicated by many variables in protocol, analysis, and interpretation. It is therefore indicated for only four situations.[1208] The first is to support an investigation of an outbreak of disease or infections when environmental reservoirs or fomites are implicated epidemiologically in disease transmission.[161, 1209, 1210] It is important that such culturing be supported by epidemiologic data. Environmental sampling, as with all laboratory testing, should not be conducted if there is no plan for interpreting and acting on the results obtained.[11, 1211, 1212] Linking microorganisms from environmental samples with clinical isolates by molecular epidemiology is crucial whenever it is possible to do so.

The second situation for which environmental sampling may be warranted is in research. Well-designed and controlled experimental methods and approaches can provide new information about the spread of health-care–associated diseases.[126, 129] A classic example is the study of environmental microbial

contamination that compared health-care–associated infection rates in an old hospital and a new facility before and shortly after occupancy.[947]

The third indication for sampling is to monitor a potentially hazardous environmental condition, confirm the presence of a hazardous chemical or biological agent, and validate the successful abatement of the hazard. This type of sampling can be used to: a) detect bioaerosols released from the operation of health-care equipment (e.g., an ultrasonic cleaner) and determine the success of repairs in containing the hazard,[1213] b) detect the release of an agent of bioterrorism in an indoor environmental setting and determine its successful removal or inactivation, and c) sample for industrial hygiene or safety purposes (e.g., monitoring a "sick building").

The fourth indication is for quality assurance to evaluate the effects of a change in infection-control practice or to ensure that equipment or systems perform according to specifications and expected outcomes. Any sampling for quality-assurance purposes must follow sound sampling protocols and address confounding factors through the use of properly selected controls. Results from a single environmental sample are difficult to interpret in the absence of a frame of reference or perspective. Evaluations of a change in infection-control practice are based on the assumption that the effect will be measured over a finite period, usually of short duration. Conducting quality-assurance sampling on an extended basis, especially in the absence of an adverse outcome, is usually unjustified. A possible exception might be the use of air sampling during major construction periods to qualitatively detect breaks in environmental infection-control measures. In one study, which began as part of an investigation of an outbreak of health-care–associated aspergillosis, airborne concentrations of *Aspergillus* spores were measured in efforts to evaluate the effectiveness of sealing hospital doors and windows during a period of construction of a nearby building.[50] Other examples of sampling for quality-assurance purposes may include commissioning newly constructed space in special care areas (i.e., ORs and units for immunosuppressed patients) or assessing a change in housekeeping practice. However, the only types of routine environmental microbiologic sampling recommended as part of a quality-assurance program are a) the biological monitoring of sterilization processes by using bacterial spores[1214] and b) the monthly culturing of water used in hemodialysis applications and for the final dialysate use dilution. Some experts also advocate periodic environmental sampling to evaluate the microbial/particulate quality for regular maintenance of the air handling system (e.g., filters) and to verify that the components of the system meet manufacturer's specifications (A. Streifel, University of Minnesota, 2000). Certain equipment in health-care settings (e.g., biological safety cabinets) may also be monitored with air flow and particulate sampling to determine performance or as part of adherence to a certification program; results can then be compared with a predetermined standard of performance. These measurements, however, usually do not require microbiologic testing.

2. Air Sampling

Biological contaminants occur in the air as aerosols and may include bacteria, fungi, viruses, and pollens.[1215, 1216] Aerosols are characterized as solid or liquid particles suspended in air. Talking for 5 minutes and coughing each can produce 3,000 droplet nuclei; sneezing can generate approximately 40,000 droplets which then evaporate to particles in the size range of 0.5–12 μm.[137, 1217] Particles in a biological aerosol usually vary in size from <1 μm to ≥50 μm. These particles may consist of a single, unattached organism or may occur in the form of clumps composed of a number of bacteria. Clumps can also include dust and dried organic or inorganic material. Vegetative forms of bacterial cells and viruses may be present in the air in a lesser number than bacterial spores or fungal spores. Factors that determine the survival of microorganisms within a bioaerosol include a) the suspending medium, b) temperature, c) relative humidity, d) oxygen sensitivity, and e) exposure to UV or electromagnetic radiation.[1215] Many vegetative cells will not survive for lengthy periods of time in the air unless the

relative humidity and other factors are favorable for survival and the organism is enclosed within some protective cover (e.g., dried organic or inorganic matter).[1216] Pathogens that resist drying (e.g., *Staphylococcus* spp., *Streptococcus* spp., and fungal spores) can survive for long periods and can be carried considerable distances via air and still remain viable. They may also settle on surfaces and become airborne again as secondary aerosols during certain activities (e.g., sweeping and bed making).[1216, 1218]

Microbiologic air sampling is used as needed to determine the numbers and types of microorganisms, or particulates, in indoor air.[289] Air sampling for quality control is, however, problematic because of lack of uniform air-quality standards. Although airborne spores of *Aspergillus* spp. can pose a risk for neutropenic patients, the critical number (i.e., action level) of these spores above which outbreaks of aspergillosis would be expected to occur has not been defined. Health-care professionals considering the use of air sampling should keep in mind that the results represent indoor air quality at singular points in time, and these may be affected by a variety of factors, including a) indoor traffic, b) visitors entering the facility, c) temperature, d) time of day or year, e) relative humidity, f) relative concentration of particles or organisms, and g) the performance of the air-handling system components. To be meaningful, air-sampling results must be compared with those obtained from other defined areas, conditions, or time periods.

Several preliminary concerns must be addressed when designing a microbiologic air sampling strategy (Box 13). Because the amount of particulate material and bacteria retained in the respiratory system is largely dependent on the size of the inhaled particles, particle size should be determined when studying airborne microorganisms and their relation to respiratory infections. Particles >5 µm are efficiently trapped in the upper respiratory tract and are removed primarily by ciliary action.[1219] Particles ≤5 µm in diameter reach the lung, but the greatest retention in the alveoli is of particles 1–2 µm in diameter.[1220–1222]

Box 13. Preliminary concerns for conducting air sampling

- **Consider the possible characteristics and conditions of the aerosol, including size range of particles, relative amount of inert material, concentration of microorganisms, and environmental factors.**
- **Determine the type of sampling instruments, sampling time, and duration of the sampling program.**
- **Determine the number of samples to be taken.**
- **Ensure that adequate equipment and supplies are available.**
- **Determine the method of assay that will ensure optimal recovery of microorganisms.**
- **Select a laboratory that will provide proper microbiologic support.**
- **Ensure that samples can be refrigerated if they cannot be assayed in the laboratory promptly.**

Bacteria, fungi, and particulates in air can be identified and quantified with the same methods and equipment (Table 23). The basic methods include a) impingement in liquids, b) impaction on solid surfaces, c) sedimentation, d) filtration, e) centrifugation, f) electrostatic precipitation, and g) thermal precipitation.[1218] Of these, impingement in liquids, impaction on solid surfaces, and sedimentation (on settle plates) have been used for various air-sampling purposes in health-care settings.[289]

Several instruments are available for sampling airborne bacteria and fungi (Box 14). Some of the samplers are self-contained units requiring only a power supply and the appropriate collecting medium, but most require additional auxiliary equipment (e.g., a vacuum pump and an airflow measuring device [i.e., a flowmeter or anemometer]). Sedimentation or depositional methods use settle plates and

therefore need no special instruments or equipment. Selection of an instrument for air sampling requires a clear understanding of the type of information desired and the particular determinations that must be made (Box 14). Information may be needed regarding a) one particular organism or all organisms that may be present in the air, b) the concentration of viable particles or of viable organisms, c) the change in concentration with time, and d) the size distribution of the collected particles. Before sampling begins, decisions should be made regarding whether the results are to be qualitative or quantitative. Comparing quantities of airborne microorganisms to those of outdoor air is also standard operating procedure. Infection-control professionals, hospital epidemiologists, industrial hygienists, and laboratory supervisors, as part of a multidisciplinary team, should discuss the potential need for microbial air sampling to determine if the capacity and expertise to conduct such sampling exists within the facility and when it is appropriate to enlist the services of an environmental microbiologist consultant.

Table 23. Air sampling methods and examples of equipment*

Method	Principle	Suitable for measuring:	Collection media or surface	Rate of collection (L/min.)	Auxilliary equipment needed+	Points to consider	Prototype samplers§
Impingement in liquids	Air drawn through a small jet and directed against a liquid surface	Viable organisms, and concentration over time. Example use: sampling water aerosols to *Legionella* spp.	Buffered gelatin, tryptose saline, peptone, nutrient broth	12.5	Yes	Antifoaming agent may be needed. Ambient temperature and humidity will influence length of collection time	Chemical Corps. All Glass Impinger (AGI)
Impaction on solid surfaces	Air drawn into the sampler; particles deposited on a dry surface	Viable particles; viable organisms (on non-nutrient surfaces, limited to organisms that resist drying and spores); size measurement, and concentration over time. Example use: sampling air for *Aspergillus* spp., fungal spores	Dry surface, coated surfaces, and agar	28 (sieve) 30–800 (slit)	Yes	Available as sieve impactors or slit impactors. Sieve impactors can be set up to measure particle size. Slit impactors have a rotating support stage for agar plates to allow for measurement of concentration over time.	Andersen Air Sampler (sieve impactor); TDL, Cassella MK-2 (slit impactors)
Sedimentation	Particles and micro-organisms settle onto surfaces via gravity	Viable particles. Example uses: sampling air for bacteria in the vicinity of and during a medical procedure; general measurements of microbial air quality.	Nutrient media (agars) on plates or slides	–	No	Simple and inexpensive; best suited for qualitative sampling; significant airborne fungal spores are too buoyant to settle efficiently for collection using this method.	Settle plates

Method	Principle	Suitable for measuring:	Collection media or surface	Rate of collection (L/min.)	Auxilliary equipment needed+	Points to consider	Prototype samplers§
Filtration	Air drawn through a filter unit; particles trapped; 0.2 µm pore size	Viable particles; viable organisms (on non-nutrient surfaces, limited to spores and organisms that resist drying); concentration over time. Example use: air sampling for *Aspergillus* spp., fungal spores, and dust	Paper, cellulose, glass wool, gelatin foam, and membrane filters	1–50	Yes	Filter must be agitated first in rinse fluid to remove and disperse trapped micro-organisms; rinse fluid is assayed; used more for sampling dust and chemicals.	–
Centrifugation	Aerosols subjected to centrifugal force; particles impacted onto a solid surface	Viable particles; viable organisms (on non-nutrient surfaces, limited to spores and organisms that resist drying); concentration over time. Example use: air sampling for *Aspergillus* spp., and fungal spores	Coated glass or plastic slides, and agar surfaces	40–50	Yes	Calibration is difficult and is done only by the factory; relative comparison of airborne contamination is its general use.	Biotest RCS Plus
Electrostatic precipitation	Air drawn over an electro-statically charged surface; particles become charged	Viable particles; viable organisms (on non-nutrient surfaces, limited to spores and organisms that resist drying); concentration over time	Solid collecting surfaces (glass, and agar)	85	Yes	High volume sampling rate, but equipment is complex and must be handled carefully; not practical for use in health-care settings.	–
Thermal precipitation	Air drawn over a thermal gradient; particles repelled from hot surfaces, settle on colder surfaces	Size measurements	Glass coverslip, and electron microscope grid	0.003–0.4	Yes	Determine particle size by direct observation; not frequently used because of complex adjustments and low sampling rates.	–

* Material in this table is compiled from references 289, 1218, 1223, and 1224.
+ Most samplers require a flow meter or anemometer and a vacuum source as auxiliary equipment.
§ Trade names listed are for identification purposes only and are not intended as endorsements by the U.S. Public Health Service.

Box 14. Selecting an air sampling device*

The following factors must be considered when choosing an air sampling instrument:

- **Viability and type of the organism to be sampled**
- **Compatibility with the selected method of analysis**
- **Sensitivity of particles to sampling**
- **Assumed concentrations and particle size**
- **Whether airborne clumps must be broken (i.e., total viable organism count vs. particle count)**
- **Volume of air to be sampled and length of time sampler is to be continuously operated**
- **Background contamination**
- **Ambient conditions**
- **Sampler collection efficiency**
- **Effort and skill required to operate sampler**
- **Availability and cost of sampler, plus back-up samplers in case of equipment malfunction**
- **Availability of auxiliary equipment and utilities (e.g., vacuum pumps, electricity, and water)**

* Material in this box is compiled from reference 1218.

Liquid impinger and solid impactor samplers are the most practical for sampling bacteria, particles, and fungal spores, because they can sample large volumes of air in relatively short periods of time.[289] Solid impactor units are available as either "slit" or "sieve" designs. Slit impactors use a rotating disc as support for the collecting surface, which allows determinations of concentration over time. Sieve impactors commonly use stages with calibrated holes of different diameters. Some impactor-type samplers use centrifugal force to impact particles onto agar surfaces. The interior of either device must be made sterile to avoid inadvertent contamination from the sampler. Results obtained from either sampling device can be expressed as organisms or particles per unit volume of air (CFU/m^3).

Sampling for bacteria requires special attention, because bacteria may be present as individual organisms, as clumps, or mixed with or adhering to dust or covered with a protective coating of dried organic or inorganic substances. Reports of bacterial concentrations determined by air sampling therefore must indicate whether the results represent individual organisms or particles bearing multiple cells. Certain types of samplers (e.g., liquid impingers) will completely or partially disintegrate clumps and large particles; the sampling result will therefore reflect the total number of individual organisms present in the air.

The task of sizing a bioaerosol is simplified through the use of sieves or slit impactors because these samplers will separate the particles and microorganisms into size ranges as the sample is collected. These samplers must, however, be calibrated first by sampling aerosols under similar use conditions.[1225]

The use of settle plates (i.e., the sedimentation or depositional method) is not recommended when sampling air for fungal spores, because single spores can remain suspended in air indefinitely.[289] Settle plates have been used mainly to sample for particulates and bacteria either in research studies or during epidemiologic investigations.[161, 1226–1229] Results of sedimentation sampling are typically expressed as numbers of viable particles or viable bacteria per unit area per the duration of sampling time (i.e., CFU/area/time); this method can not quantify the volume of air sampled. Because the survival of microorganisms during air sampling is inversely proportional to the velocity at which the air is taken into the sampler,[1215] one advantage of using a settle plate is its reliance on gravity to bring organisms and particles into contact with its surface, thus enhancing the potential for optimal survival of collected organisms. This process, however, takes several hours to complete and may be impractical for some situations.

Air samplers are designed to meet differing measurement requirements. Some samplers are better suited for one form of measurement than others. No one type of sampler and assay procedure can be used to collect and enumerate 100% of airborne organisms. The sampler and/or sampling method chosen should, however, have an adequate sampling rate to collect a sufficient number of particles in a reasonable time period so that a representative sample of air is obtained for biological analysis. Newer analytical techniques for assaying air samples include PCR methods and enzyme-linked immunosorbent assays (ELISAs).

3. Water Sampling

A detailed discussion of the principles and practices of water sampling has been published.[945] Water sampling in health-care settings is used detect waterborne pathogens of clinical significance or to determine the quality of finished water in a facility's distribution system. Routine testing of the water in a health-care facility is usually not indicated, but sampling in support of outbreak investigations can help determine appropriate infection-control measures. Water-quality assessments in dialysis settings have been discussed in this guideline (see Water, Dialysis Water Quality and Dialysate, and Appendix C).

Health-care facilities that conduct water sampling should have their samples assayed in a laboratory that uses established methods and quality-assurance protocols. Water specimens are not "static specimens" at ambient temperature; potential changes in both numbers and types of microbial populations can occur during transport. Consequently, water samples should be sent to the testing laboratory cold (i.e., at approximately 39.2°F [4°C]) and testing should be done as soon as practical after collection (preferably within 24 hours).

Because most water sampling in health-care facilities involves the testing of finished water from the facility's distribution system, a reducing agent (i.e., sodium thiosulfate [$Na_2S_2O_3$]) needs to be added to neutralize residual chlorine or other halogen in the collected sample. If the water contains elevated levels of heavy metals, then a chelating agent should be added to the specimen. The minimum volume of water to be collected should be sufficient to complete any and all assays indicated; 100 mL is considered a suitable minimum volume. Sterile collection equipment should always be used.

Sampling from a tap requires flushing of the water line before sample collection. If the tap is a mixing faucet, attachments (e.g., screens and aerators) must be removed, and hot and then cold water must be run through the tap before collecting the sample.[945] If the cleanliness of the tap is questionable, disinfection with 500–600 ppm sodium hypochlorite (1:100 v/v dilution of chlorine bleach) and flushing the tap should precede sample collection. The verification code for this document is 940210.

Microorganisms in finished or treated water often are physically damaged ("stressed") to the point that growth is limited when assayed under standard conditions. Such situations lead to false-negative readings and misleading assessments of water quality. Appropriate neutralization of halogens and chelation of heavy metals are crucial to the recovery of these organisms. The choice of recovery media and incubation conditions will also affect the assay. Incubation temperatures should be closer to the ambient temperature of the water rather than at 98.6°F (37°C), and recovery media should be formulated to provide appropriate concentrations of nutrients to support organisms exhibiting less than rigorous growth.[945] High-nutrient content media (e.g., blood agar and tryptic soy agar [TSA]) may actually inhibit the growth of these damaged organisms. Reduced nutrient media (e.g., diluted peptone and R2A) are preferable for recovery of these organisms.[945]

Use of aerobic, heterotrophic plate counts allows both a qualitative and quantitative measurement for water quality. If bacterial counts in water are expected to be high in number (e.g., during waterborne outbreak investigations), assaying small quantities using pour plates or spread plates is appropriate.[945] Membrane filtration is used when low-count specimens are expected and larger sampling volumes are required ($\geq$100 mL). The sample is filtered through the membrane, and the filter is applied directly face-up onto the surface of the agar plate and incubated.

Unlike the testing of potable water supplies for coliforms (which uses standardized test and specimen collection parameters and conditions), water sampling to support epidemiologic investigations of disease outbreaks may be subjected to modifications dictated by the circumstances present in the facility. Assay methods for waterborne pathogens may also not be standardized. Therefore, control or comparison samples should be included in the experimental design. Any departure from a standard method should be fully documented and should be considered when interpreting results and developing strategies. Assay methods specific for clinically significant waterborne pathogens (e.g., *Legionella* spp., *Aeromonas* spp, *Pseudomonas* spp., and *Acinetobacter* spp.) are more complicated and costly compared with both methods used to detect coliforms and other standard indicators of water quality.

4. Environmental Surface Sampling

Routine environmental-surface sampling (e.g., surveillance cultures) in health-care settings is neither cost-effective nor warranted.[951, 1225] When indicated, surface sampling should be conducted with multidisciplinary approval in adherence to carefully considered plans of action and policy (Box 15).

Box 15. Undertaking environmental-surface sampling*

The following factors should be considered before engaging in environmental-surface sampling:

- **Background information from the literature and present activities (i.e., preliminary results from an epidemiologic investigation)**
- **Location of surfaces to be sampled**
- **Method of sample collection and the appropriate equipment for this task**
- **Number of replicate samples needed and which control or comparison samples are required**
- **Parameters of the sample assay method and whether the sampling will be qualitative, quantitative, or both**
- **An estimate of the maximum allowable microbial numbers or types on the surface(s) sampled (refer to the Spaulding classification for devices and surfaces)**
- **Some anticipation of a corrective action plan**

* The material in this box is compiled from reference 1214.

Surface sampling is used currently for research, as part of an epidemiologic investigation, or as part of a comprehensive approach for specific quality assurance purposes. As a research tool, surface sampling has been used to determine a) potential environmental reservoirs of pathogens,[564, 1230–1232] b) survival of microorganisms on surfaces,[1232, 1233] and c) the sources of the environmental contamination.[1023] Some or all of these approaches can also be used during outbreak investigations.[1232] Discussion of surface sampling of medical devices and instruments is beyond the scope of this document and is deferred to future guidelines on sterilization and disinfection issues.

Meaningful results depend on the selection of appropriate sampling and assay techniques.[1214] The media, reagents, and equipment required for surface sampling are available from any well-equipped

microbiology laboratory and laboratory supplier. For quantitative assessment of surface organisms, non-selective, nutrient-rich agar media and broth (e.g., TSA and brain-heart infusion broth [BHI] with or without 5% sheep or rabbit blood supplement) are used for the recovery of aerobic bacteria. Broth media are used with membrane-filtration techniques. Further sample work-up may require the use of selective media for the isolation and enumeration of specific groups of microorganisms. Examples of selective media are MacConkey agar (MAC [selects for gram-negative bacteria]), Cetrimide agar (selects for *Pseudomonas aeruginosa*), or Sabouraud dextrose- and malt extract agars and broths (select for fungi). Qualitative determinations of organisms from surfaces require only the use of selective or non-selective broth media.

Effective sampling of surfaces requires moisture, either already present on the surface to be sampled or via moistened swabs, sponges, wipes, agar surfaces, or membrane filters.[1214, 1234–1236] Dilution fluids and rinse fluids include various buffers or general purpose broth media (Table 24). If disinfectant residuals are expected on surfaces being sampled, specific neutralizer chemicals should be used in both the growth media and the dilution or rinse fluids. Lists of the neutralizers, the target disinfectant active ingredients, and the use concentrations have been published.[1214, 1237] Alternatively, instead of adding neutralizing chemicals to existing culture media (or if the chemical nature of the disinfectant residuals is unknown), the use of either a) commercially available media including a variety of specific and non-specific neutralizers or b) double-strength broth media will facilitate optimal recovery of microorganisms. The inclusion of appropriate control specimens should be included to rule out both residual antimicrobial activity from surface disinfectants and potential toxicity caused by the presence of neutralizer chemicals carried over into the assay system.[1214]

Table 24. Examples of eluents and diluents for environmental-surface sampling* +

Solutions	Concentration in water
Ringer	¼ strength
Peptone water	0.1%–1.0%
Buffered peptone water	0.067 M phosphate, 0.43% NaCl, 0.1% peptone
Phosphate-buffered saline	0.02 M phosphate, 0.9% NaCl
Sodium chloride (NaCl)	0.25%–0.9%
Calgon Ringer§	¼ strength
Thiosulfate Ringer¶	¼ strength
Water	–
Tryptic soy broth (TSB)	–
Brain-heart infusion broth (BHI) supplemented with 0.5% beef extract	–

* Material in this table is compiled from references 1214 and 1238.

\+ A surfactant (e.g., polysorbate [i.e., Tween® 80]) may be added to eluents and diluents. A concentration ranging from 0.01%–0.1% is generally used, depending on the specific application. Foaming may occur during use.

§ This solution is used for dissolution of calcium alginate swabs.

¶ This solution is used for neutralization of residual chlorine.

Several methods can be used for collecting environmental surface samples (Table 25). Specific step-by-step discussions of each of the methods have been published.[1214, 1239] For best results, all methods should incorporate aseptic techniques, sterile equipment, and sterile recovery media.

Table 25. Methods of environmental-surface sampling

Method	Suitable for appropriate surface(s)	Assay technique	Procedural notes	Points of interpretation	Available standards	References
Sample/rinse Moistened swab/rinse	Non-absorbent surfaces, corners, crevices, devices, and instruments	Dilutions; qualitative or quantitative assays	Assay multiple measures areas or devices with separate swabs	Report results per measured areas or if assaying an object, per the entire sample site	**YES** – food industry; **NO** – heath care	1214, 1239–1242
Moistened sponge/rinse	Large areas and housekeeping surfaces (e.g., floors or walls)	Dilutions; qualitative or quantitative assays	Vigorously rub a sterile sponge over the surface	Report results per measured area	**YES** – food industry; **NO** – health care	1214, 1239–1242
Moistened wipe/rinse	Large areas and housekeeping surfaces (e.g., countertops)	Dilutions; qualitative or quantitative assays	Use a sterile wipe	Report results per measured area	**YES** – food industry; **NO** – health care	1214, 1239–1242
Direct immersion	Small items capable of being immersed	Dilutions; qualitative or quantitative assays	Use membrane filtration if rinse volume is large and anticipated microbiological concentration is low	Report results per item	**NO**	1214
Containment	Interior surfaces of containers, tubes, or bottles	Dilutions; qualitative or quantitative assays	Use membrane filtration if rinse volume is large	Evaluate both the types and numbers of microorganisms	**YES** – food and industrial applications for containers prior to fill	1214
RODAC*	Previously cleaned and sanitized flat, non-absorbent surfaces; not suitable for irregular surfaces	Direct assay	Overgrowth occurs if used on heavily contaminated surfaces; use neutralizers in the agar if surface disinfectant residuals are present	Provides direct, quantitative results; use a minimum of 15 plates per an average hospital room	**NO**	1214, 1237, 1239, 1243, 1244

* RODAC stands for "replicate organism direct agar contact."

Sample/rinse methods are frequently chosen because of their versatility. However, these sampling methods are the most prone to errors caused by manipulation of the swab, gauze pad, or sponge.[1238] Additionally, no microbiocidal or microbiostatic agents should be present in any of these items when used for sampling.[1238] Each of the rinse methods requires effective elution of microorganisms from the item used to sample the surface. Thorough mixing of the rinse fluids after elution (e.g., via manual or mechanical mixing using a vortex mixer, shaking with or without glass beads, and ultrasonic bath) will help to remove and suspend material from the sampling device and break up clumps of organisms for a more accurate count.[1238] In some instances, the item used to sample the surface (e.g., gauze pad and sponge) may be immersed in the rinse fluids in a sterile bag and subjected to stomaching.[1238] This technique, however, is suitable only for soft or absorbent items that will not puncture the bag during the elution process.

If sampling is conducted as part of an epidemiologic investigation of a disease outbreak, identification of isolates to species level is mandatory, and characterization beyond the species level is preferred.[1214] When interpreting the results of the sampling, the expected degree of microbial contamination

associated with the various categories of surfaces in the Spaulding classification must be considered. Environmental surfaces should be visibly clean; recognized pathogens in numbers sufficient to result in secondary transfer to other animate or inanimate surfaces should be absent from the surface being sampled.1214 Although the interpretation of a sample with positive microbial growth is self-evident, an environmental surface sample, especially that obtained from housekeeping surfaces, that shows no growth does not represent a "sterile" surface. Sensitivities of the sampling and assay methods (i.e., level of detection) must be taken into account when no-growth samples are encountered. Properly collected control samples will help rule out extraneous contamination of the surface sample.