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SECTION 3

Basic Principles for Infection Control

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CHAPTER 11

Infection Control Programs for Dogs and Cats

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KEY POINTS

- A hospital infection control program consists of infectious disease control personnel, a written protocol, training, and documentation. The aim of such a program is to reduce the incidence of hospital-acquired infections among patients, staff, and visitors to a small animal hospital.
- The infection control program describes the requirement for practices that optimize hygiene such as hand washing, the use of protective clothing, cleaning and disinfection, and appropriate disposal of infectious agents. The protocol can also be used to educate staff about specific transmission precautions for infectious diseases seen within a practice.
- This chapter describes the infection control program and important components of an infection control protocol, including the advantages and disadvantages of various methods of sterilization, disinfection, and antisepsis used in small animal hospitals.
- Information in this chapter also has relevance to infection control in shelters and in breeding and boarding facilities.

INTRODUCTION

The primary role of an infection control program is to reduce the incidence of hospital-acquired infections (HAIs) by patients, staff, and visitors to a small animal hospital. Infection control programs in veterinary hospitals have largely evolved from evidence and protocols from the human health care setting, together with our understanding of transmission pathways for veterinary pathogens. In recent years, several factors have led to an increased rate of adoption of infection control programs by veterinary hospitals, such as an increased prevalence of multidrug-resistant bacterial infections among small animal patients, the appearance of more studies that support the common occurrence of transmission of hospital-associated and zoonotic pathogens in the veterinary setting, the requirement for an infection control program in veterinary teaching hospitals for accreditation purposes, and increased scrutiny of hospital-associated and zoonotic infections to protect hospitals against litigation.

Infection Control Programs

Every small animal hospital should have an infection control program. At a minimum, this should consist of an infectious disease control officer, a written infection control protocol, regular training of staff, and documents that record all training and surveillance efforts. In large hospitals, formation of an infectious disease control committee may be required. In this situation, the infectious disease control committee could consist of an internist or criticalist with an interest or training in infectious diseases, a veterinary clinical microbiologist, a nursing supervisor, a safety officer, the hospital administrator, and the hospital director. There should be adequate personnel and communication so that proper coverage is maintained when one individual is absent or unavailable. All personnel who work in a veterinary hospital, as well as visitors, should be familiar with the infectious disease control personnel as well as procedures and policies listed in the infection control protocol. Documentation of training should be maintained for all hospital personnel.

The Hospital Infection Control Protocol

The objective of a hospital infection control protocol is to provide a standard procedure for the control of infectious diseases in the hospital, in order to minimize animal-to-animal, animal-to-human, and human-to-animal transmission of pathogens. Adherence to the infection control protocol can also reduce transmission of infectious agents between personnel through increased hand washing and reduction of fomite contamination. The infection control protocol is a legal document and should

be regularly updated by a designated hospital infection control officer or committee. Each hospital should develop a protocol that is tailored to address specific practice requirements and the hospital design, purpose, and equipment used. Additional special precautions may need to be described for hospitals that see avian and exotic pet animal species, and in geographic locations where serious zoonotic diseases such as rabies and plague are endemic (see Chapters 13 and 55).

The infection control protocol details practices that optimize hygiene such as hand washing, the use of protective clothing, cleaning and disinfection, and appropriate disposal of infectious agents. Specific infectious disease control procedures to be followed in different areas of the hospital (radiology, surgery, the intensive care unit, isolation, wards) can be included, as well as policies on antimicrobial use. The protocol can also be used to educate staff about routes of transmission, the potential for zoonotic transmission, specific transmission precautions for infectious diseases that are seen within the practice, and immunization requirements, such as those for rabies (see Chapter 13).

Standard Precautions

All animals that enter a veterinary hospital are potential carriers of pathogens that may be spread to other animals or people. Animals may be colonized with multidrug-resistant bacteria, often without showing evidence of disease due to these organisms. Pet owners may carry these organisms on their hands and clothing. Standard precautions such as hand hygiene and the wearing of routine protective clothing can minimize the spread of these bacteria around the hospital and to immunocompromised animals.

Hand Hygiene

Frequent and proper hand and wrist washing to remove transient flora on the hands has been proven as the most important component for prevention of the spread of infectious diseases in human hospitals.[1] Signs that outline proper hand-washing technique, including the use of paper towels to turn off the faucet, posted adjacent to basins around the hospital can improve compliance among staff. Online videos that demonstrate proper hand-washing technique are available for educational purposes.[2] Guidelines for hand washing are shown in Box 11-1. Antibacterial soap should be used, and all surfaces of the hands should be rubbed together, which should include the backs of the hands, between the fingers, and under the fingernails, for a total hand-washing time of at least 20 seconds. In order to prevent chapped skin, which can harbor bacteria, water used for hand washing should not be too hot, and hand lotion should be applied regularly. Behaviors such as keeping fingernails short, avoidance of artificial and/or polished fingernails or hand jewelry, or wearing jewelry on a chain around the neck instead of on the hand can be encouraged. Meticulous hand hygiene is particularly important for personnel who work frequently with immunocompromised animals, such as emergency and critical care personnel. The use of touch-free taps and paper towel dispensers can also reduce transmission of bacteria during hand washing.

Alcohol-based hand sanitizers are a more convenient form of sanitization. These can be provided in multiple locations around a hospital, or travel-sized bottles can be carried in a coat pocket. At least one to two full pumps or a 3-cm diameter pool of the product should be dispensed onto one palm. All surfaces of the hands and wrists should be rubbed with the product until it has dried. Use of soap and water, rather than hand sanitizer, is recommended when there is gross contamination with organic matter, or when exposure to alcohol-resistant pathogens such as *Clostridium* spp. spores or parvovirus might have occurred. However, the availability of hand sanitizers improves compliance in busy hospital situations, is associated with lower rates of dermatitis than medicated soaps, and is recommended for routine hand sanitation in human health care settings by the Centers for Disease Control and Prevention (CDC) and the World Health Organization (WHO).[3,4]

The use of gloves prevents contamination of the hands with microorganisms, prevents exposure to bloodborne pathogens, and reduces the risk of transmission of microorganisms from personnel to animals. However, gloves are not a substitute for proper hand hygiene. Guidelines for wearing disposable gloves are shown in Box 11-1. Gloves should be promptly removed after use, before other surfaces are touched, and hands should then be washed. If a glove is torn or punctured, it should be removed and replaced as soon as possible.

BOX 11-1

Guidelines for Hand Washing and the Wearing of Disposable Gloves

Situations That Require Hand Washing

Immediately before handling a patient
Immediately after handling a patient
Immediately before and after procedures that involve nonintact skin
Before and after gloves are worn for procedures
After blood, body fluids, secretions, excretions, or contaminated items are touched
After cages are cleaned
Before and after eating, smoking, or leaving the hospital
After going to the restroom

Situations When Disposable Gloves Should Be Worn

Contact with broken skin and bodily fluids (blood, urine, and feces)
Handling of disinfectants
Handling of animals with suspected or known infectious diseases
Handling of all animals for immunocompromised people

Hospital Attire

Nonsurgical Areas

Staff should be encouraged to wear dedicated hospital attire that is not worn elsewhere, so that hospital pathogens are not transported to and from locations outside the hospital. At the minimum, protective clothing such as clean laboratory coats or hospital scrubs and closed-toe shoes must be worn in nonsurgical areas. Sleeves must be short enough or rolled up to expose the wrists, and laboratory coats changed whenever gross soiling occurs. In the absence of gross soiling, coats should be changed daily. If neckties are worn, they must be secured in place by an outer layer of clothing or a tie pin so that they cannot be contaminated as a result of contact with patients or environmental

TABLE 11-1

Transmission Precautions for Selected Infectious Diseases of Dogs and Cats

	Infectious agent	Specific precautions
Airborne	*Mycobacterium tuberculosis* *Yersinia pestis* *Francisella tularensis*	Isolation, preferably in negative pressure facility Properly fitted N95 respirator masks Contact precautions
Droplet	Canine and feline transmissible respiratory disease pathogens (e.g., canine distemper virus, canine respiratory coronavirus, feline herpesvirus-1, feline calicivirus)	Isolation Space animals at least 4 feet apart Contact precautions
Contact	Multidrug-resistant bacteria Dermatophytes *Leptospira* spp. *Salmonella* spp. Parvoviruses	Warning signage Gown, gloves, dedicated equipment Isolation for select pathogens (see Box 11-2) Limitation of movement Standard hand hygiene precautions Proper cleaning, disinfection, and disposal of medical waste

surfaces and act as fomites, as has been shown to occur in human hospital environments.[5] Long hair must be tied back so that it does not drape on animals and hospital surfaces. Face shields and a clean gown should be worn during procedures that are likely to generate splashes or sprays of blood and body fluids. Gowns must be made of impervious material and tied on securely and correctly. Soiled gowns must be removed as soon as they are no longer required and face shields cleaned.

Surgical Areas

Dedicated operating room attire should be worn in surgical areas and should be changed after gross soilage and when leaving the operating room for the day. Caps and masks should be worn and hands thoroughly washed on entry to the operating room. There is no evidence that dedicated footwear or foot protection should be worn for prevention of surgical site infections in human patients, and major human guidelines do not advocate that footwear be addressed.[6] Protective wear should be removed whenever staff members leave the operating room. Traffic in and out of the operating room should be limited to the minimum required for patient care.

General Animal Handling Precautions

All animals seen at a veterinary hospital should undergo a history and physical examination by a veterinarian to determine the likelihood and nature of any transmissible infections that might be present. Ideally, client beds, blankets, collars, and leashes should not be brought into the hospital, where they could become contaminated. Animals should always be placed in cages that have been cleaned and disinfected appropriately. Disposable thermometer sleeves should always be used on thermometers. Equipment should not be shared between animals unless it has been cleaned and disinfected. Diets that contain raw meat and bones should not be fed or stored in the hospital, because they commonly contain and can potentially transmit foodborne gastrointestinal pathogens.[7] The handling of sick animals should be minimized, unless required for patient care.

Because some infections can be transmitted through bites and scratches, staff should be educated on bite and scratch avoidance, such as the use of restraint devices, protective gloves, and warning signage on cages and medical records. Should bites or scratches occur, they should be vigorously flushed and immediately washed with water and chlorhexidine or a dilute iodophor solution, and the bite reported to appropriate officials as necessary. Deep wounds could be irrigated with pressure using a syringe without an attached needle. If a bite occurs, medical attention should be sought as soon as possible. All bites or scratches must be documented and consideration be given as to why the injury occurred, so that procedures or training to prevent future injuries can be implemented if necessary.

Consumption of food and drink should be limited to parts of a hospital where patient care and the handling of biologic specimens and medications do not occur. Food and beverages should not be left out open on benches for long periods. Microwaves used for animal care purposes should not be used to heat food intended for people.

Transmission-Based Precautions

Transmission-based precautions are instituted for selected patients that are confirmed to be or suspected to be infected or colonized with important transmissible pathogens. Transmission-based precautions are used in combination with standard precautions. In the human hospital setting, three types of transmission-based precautions have been developed—airborne, droplet, and contact precautions (Table 11-1).[8] *Airborne precautions* are used to prevent the transmission of diseases by droplet nuclei (particles <5 μm). Transmission by droplet nuclei occurs with diseases such as measles, varicella, and pulmonary tuberculosis. The precautions for human patients involve isolation in a single-bed, negative-pressure room and the wearing of high-density respirator (N95) masks. These resemble surgical masks but filter 1-μm particles with an efficiency of at least 95% and must be properly fitted. Airborne contact precautions are rarely necessary in hospitals that treat only dogs and cats, but could be considered when animals suspected to have pneumonic plague, tularemia, or *Mycobacterium tuberculosis* infection. *Droplet precautions* are used to prevent transmission by large-particle aerosols and do not require a negative-pressure room. Droplet precautions apply to dogs and cats with transmissible respiratory disease. *Contact precautions* are indicated

for animals with infections that can be transmitted by direct contact with the patient or through fomite contact.

If it is known in advance that an animal with a suspected or known transmissible disease is to be seen at the hospital, arrangements should be made to have the pet owner take the animal from the parking area directly to an examination room, so that the animal does not contaminate the waiting area. After the animal has been examined, appropriate signage should be placed on the door of the examination room to prevent use until it has been properly cleaned and disinfected.

If hospitalization is required, animals with known or suspected transmissible disease must be admitted either to an isolation ward or to regular hospital cages with handling precautions, based on the pathogen suspected. Immediately after hospitalization, the name of the suspected or known pathogen should be posted on the cage or run, together with a handling precautions notice. Animals that require contact precautions should be placed in cages away from other animals in the ward and should not be moved from one cage to another, unless there is a medical need. Animals with transmissible respiratory disease should be placed in isolation and separated both horizontally and vertically from other patients by at least 4 feet. Gloves and a gown should be put on before the patient is handled, and then removed and disposed of immediately afterward. Hands should be washed after the gloves are removed. Additional items of personal protective equipment (masks, gowns, gloves, booties) may be required in some circumstances. Soiled linen and equipment should be handled, transported, and processed in a manner that prevents skin and mucous membrane exposures and contamination of clothing and that avoids transfer of microorganisms to other patients and environments. All equipment that contacts the patient (scales, examination tables, stethoscope heads, floor) should be cleaned and disinfected immediately after use. Medications and fluids from these patients should not be returned to the hospital pharmacy. When possible, personnel who handle these animals should not work with other immunosuppressed animals in the hospital, or they should work with the infectious disease cases last. If animals with contact precautions must be moved within the hospital, personnel should ensure that precautions are maintained throughout and that equipment and environmental surfaces that come into contact with the patient are properly disinfected and/or disposed of. The cleaning and bandaging of wounds infected with multidrug-resistant bacteria should be conducted in low-traffic areas that can be properly cleaned and disinfected.

The owners of dogs and cats that have transmissible diseases should be provided with general information regarding the risk of disease transmission to in-contact animals and people that includes the mode of transmission, duration of organism shedding, and if there are special implications for young children or other immunosuppressed individuals. If dogs and cats are diagnosed with a zoonotic disease, the owners should be notified without delay. The owners should be told to see their physicians if they become unwell or, in some circumstances, immediately, and to advise a physician of the potential exposure.

Isolation

In contrast to human hospitals where patients can be more readily isolated in single-bed rooms or cubicles, isolation of veterinary patients can be more difficult because of the close proximity of one animal to another. Floor contamination with secretions and excretions can also occur more readily. Isolation rooms are available in many veterinary hospitals, but they may be poorly visible and/or accessible and may not provide access to an oxygen source or be amenable to intensive monitoring and care. For some animals (especially puppies and kittens) suspected to have a transmissible disease, housing in a general ward or ICU area with as much physical and procedural separation as possible, and with strict infection control practices, may be acceptable (if perhaps not optimal). Once a diagnosis is confirmed, the animal should be moved immediately to isolation whenever possible. Patients chosen for strict isolation vary based on the specific situation and facilities available, but suggestions are provided in Box 11-2.

Only the individuals directly involved in the care of the patient should enter isolation. Pet owners should not be allowed into the isolation ward. No equipment used outside isolation (pens, thermometers, stethoscopes, cell phones) should be brought into isolation. Laboratory coats should be removed, and personnel must put on protective wear such as a disposable gown, gloves, and booties when entering the isolation ward. Face protection may also be required, depending on the situation. A notice that outlines the required precautions should be posted on the door of isolation. Protective clothing should be removed before leaving isolation, and hands should be washed. Once a patient has been discharged, the room should be properly disinfected.

Handling of Potentially Infectious Materials and Waste

The risk of human infection from patient blood and body fluids in small animal hospitals is clearly lower than that in human hospitals. However, a number of zoonotic pathogens have the potential to be spread from dogs or cats to humans through contact with medical waste, and a number of emerging zoonotic infectious agents are bloodborne. Examples include *Bartonella* spp., *Anaplasma phagocytophilum, Brucella* spp., and hemoplasmas. Proper handling of blood and body fluids also has the potential to protect staff against as-yet-unrecognized zoonotic pathogens. Improper disposal of medical waste in veterinary hospitals risks injuring others who handle the waste and has the potential to result in serious penalties or fines.

Potentially contaminated waste should be disposed of in an approved plastic bag in a container labeled on all exposed sides

BOX 11-2

Examples of Infectious Agents of Dogs and Cats for Which Strict Isolation Is Indicated

Salmonella spp.
Francisella tularensis
Yersinia pestis
Mycobacterium tuberculosis or *Mycobacterium bovis*
Microsporum canis
Rabies virus
Enteric viruses, such as parvoviruses
Canine transmissible respiratory disease pathogens (*Bordetella*, canine distemper virus, influenza viruses, canine respiratory coronavirus, canine adenovirus, canine parainfluenza virus)
Feline upper respiratory tract disease pathogens (feline herpesvirus-1, feline calicivirus, influenza virus, *Chlamydia*)

with "biohazardous waste." Blood-soaked materials, infected materials, and empty fluid bags should all be placed in biohazardous waste containers. Care should be taken not to contaminate the outside of the container during disposal. The lid of the biohazardous waste container must close properly. Blood and body fluids should be inactivated with an appropriate disinfectant (e.g., bleach or accelerated hydrogen peroxide) and allowed to stand for 10 minutes before disposal. Disposal regulations may vary depending on local laws, but liquid waste should not be disposed of into storm drains.

Specimens for laboratory testing that are collected from a patient with suspected zoonotic infectious diseases should be placed in an outer plastic bag, taking care not to contaminate the outside of the plastic bag, and labeled with an appropriate warning label. Fecal material should be picked up using a tongue depressor while wearing gloves, and placed in a sealed plastic cup and clearly labeled. Urine specimens should be submitted in a sealed container.

Sharps Handling

Sharps handling practices receive considerable attention in human medicine because of the risk of transmission of various bloodborne pathogens. Although the risks are less in veterinary medicine, significant injury or illness can follow sharps injuries, such as transmission of infectious agents from the patient, allergic or inflammatory reactions from exposure to medication, and inoculation of opportunistic pathogens from the injured person's own skin microflora. Care should be taken to prevent injuries when needles, scalpels, and other sharp instruments are used, cleaned, or disposed of. Used needles should never be recapped, and they should not be removed from the barrel of a disposable syringe. Personnel should be instructed not to walk with uncapped needles and not to hold syringe or needle caps in the mouth. Used needles should not be carried in a pocket. Animals suspected of having an infectious disease that could be transmitted to humans through a needle-stick injury should be sedated before skin masses or peripheral lymph nodes are aspirated or venipuncture is performed. Contaminated sharps (slides, scalpel blades, broken glass, needles with attached syringes) must immediately be placed into an approved puncture-resistant sharps disposal biohazard container. Personnel who perform necropsy examinations should take care to use sharp knives in the proper manner and to avoid rushed situations. If a contaminated sharps injury occurs, medical attention should be sought immediately if necessary.

Hospital Cleaning, Disinfection, and Sterilization

Definitions

Sterilization refers to complete elimination of all microbes, including bacterial spores, and is accomplished in hospital settings using processes such as pressurized steam, dry heat, ethylene oxide gas, or liquid chemicals.

Disinfection is the process that eliminates many or all microbes from inanimate objects, but not bacterial spores. Factors that influence the efficacy of disinfection include the type of microorganism present, their number, the amount and type of organic matter present, the presence of biofilms, and the porosity of the surface to be disinfected. Some disinfectants kill spores at high concentrations and with prolonged exposure times. These are known as *chemical sterilants*. At low concentrations and short contact times, chemical sterilants inactivate all microbes except large numbers of bacterial spores and are known as *high-level disinfectants*. *Low-level disinfectants* inactivate most vegetative bacteria, some fungi, and enveloped viruses, but not bacterial spores. *Intermediate-level disinfectants* inactivate mycobacteria, vegetative bacteria, most viruses, and most fungi (Tables 11-2 and 11-3).

Antisepsis is the process that reduces the number of microbes from *living tissue and skin*. Disinfectants are rarely used for skin antisepsis because they can injure tissues and skin. *Sanitation* is the reduction in the number of microorganisms *on a surface* to a safe level. *Germicides* are agents that inactivate microorganisms and include disinfectants, antiseptics, and sanitizers.

TABLE 11-2

Characteristics of Selected Disinfectants

Disinfectant Category	Activity in the Presence of Organic Matter	Advantages	Disadvantages	Precautions	Comments
Alcohols: Ethyl alcohol, isopropyl alcohol	Rapidly inactivated	Fast-acting No residue Relatively nontoxic	Rapid evaporation	Flammable	Not appropriate for routine environmental disinfection Primarily used as antiseptics
Aldehydes: Formaldehyde, glutaraldehyde	Good	Broad spectrum Relatively noncorrosive	Highly toxic	Irritant Carcinogenic Requires ventilation	Used as an aqueous solution or as a gas (fumigation)
Alkalis: Ammonia			Unpleasant odor Irritating	Do not mix with bleach.	Not recommended for general use
Biguanides: Chlorhexidine	Rapidly inactivated	Nontoxic	Incompatible with anionic detergents		Not appropriate for environmental disinfection Primarily used as antiseptics

Continued

TABLE 11-2

Characteristics of Selected Disinfectants—cont'd

Disinfectant Category	Activity in the Presence of Organic Matter	Advantages	Disadvantages	Precautions	Comments
Halogens: Hypochlorites (bleach)	Rapidly inactivated	Broad spectrum, sporicidal Inexpensive Can be used on food preparation surfaces	Inactivated by cationic soaps/detergents and sunlight. Frequent application required.	Corrosive Irritant May produce toxic gas when mixed with other chemicals	Used to disinfect clean environmental surfaces Only commonly available sporicidal disinfectant
Oxidizing agents	Good	Broad spectrum Environmentally friendly	Breakdown with time	Corrosive	Excellent choice for environmental disinfection
Phenolics	Good	Broad spectrum Noncorrosive Stable in storage	Toxic to cats Unpleasant odor Incompatible with cationic or nonionic detergents	Irritant	Some residual activity after drying
Quaternary ammonium compounds (QUATs)	Moderate	Stable in storage Nonirritating to skin Low toxicity Can be used on food preparation surfaces Effective at high temperatures and pH	Incompatible with anionic detergents		Commonly used primary environmental disinfectant Some residual activity after drying

Source: Modified from the Canadian Committee on Antibiotic Resistance. Infection Prevention and Control Best Practices for Small Animal Veterinary Clinics, 2008; http://www.wormsandgermsblog.com/2008/04/promo/services/infection-prevention-and-control-best-practices-for-small-animal-veterinary-clinics/. Last accessed May 15, 2012.

TABLE 11-3

Antimicrobial Spectrum of Selected Disinfectants

Agent	Alcohols	Aldehydes	Alkalis: Ammonia	Biguanides: Chlorhexidine	Halogens: Hypochlorite (Bleach)	Oxidizing Agents	Phenolics	Quaternary Ammonium Compounds
Mycoplasmas	++	++	++	++	++	++	++	+
Gram-positive bacteria	++	++	+	++	++	++	++	++
Gram-negative bacteria	++	++	+	+	++	++	++	+
Pseudomonads	++	++	+	±	++	++	++	±
Enveloped viruses	+	++	+	++	++	++	++	+
Non-enveloped viruses	–	+	±	–	++	+	±*	–
Fungal spores	±	+	+	±	+	±	+	±
Mycobacteria	+	++	+	–	+	±	++	–
Bacterial spores	–	+	±	–	++	+	–	–

++, Highly effective; +, Effective; ±, Limited activity; –, No activity.

*In general, phenols are not active against non-enveloped viruses but they do have some activity against rotaviruses. Activity against parvoviruses has not been documented.

Examples of microorganisms from each category:

Mycoplasmas: *Mycoplasma cynos*, *Mycoplasma felis*; **Gram-positive bacteria**: *Staphylococcus* spp., *Streptococcus* spp.; **Gram-negative bacteria**: *Bordetella bronchiseptica*, *Salmonella* spp.; **Pseudomonads**: *Pseudomonas aeruginosa*; **Enveloped viruses**: influenza virus, herpesvirus; **Non-enveloped viruses**: feline panleukopenia virus, canine parvovirus, feline calicivirus; **Fungal spores**: *Aspergillus* spp.; **Acid-fast bacteria**: *Mycobacterium fortuitum*; **Bacterial spores**: *Clostridium difficile*, *Clostridium perfringens*.

Source: Modified from the Canadian Committee on Antibiotic Resistance. Infection Prevention and Control Best Practices for Small Animal Veterinary Clinics, 2008; http://www.wormsandgermsblog.com/2008/04/promo/services/infection-prevention-and-control-best-practices-for-small-animal-veterinary-clinics/. Last accessed May 15, 2012.

TABLE 11-4

Recommended Conditions for Heat Sterilization

	Conditions	Time
Gravity displacement steam sterilization	121°C (250°F), 106 kPa (15 lb/in^2)* 132°C (270°F), 30 lb/in^2	30 min wrapped items, 20 min unwrapped 15 min wrapped items
High-speed prevacuum steam sterilization	132°C (270°F), 30 lb/in^2	4 min
Dry heat sterilization	160°C (320°F) 170°C (340°F)	2 hours at temperature, 3 to 3.5 hours with cooling 1 hour at temperature, 2 to 2.5 hours with cooling

*Pressure settings may vary between incubators. When possible, follow manufacturer's recommendations.

Cleaning involves the removal of all visible organic and inorganic material from objects and surfaces through the use of manual or mechanical processes and detergent or enzymatic solutions.

According to the Spaulding method of classification, items to be sterilized or disinfected can be grouped into *critical, semicritical,* and *noncritical* items.[9] Critical items enter tissue or the vascular system, or devices through which blood flows. Critical items require sterilization before they can be used. Semicritical items are items that contact mucous membranes or nonintact skin and include endoscopes and balloon dilation catheters. These generally require high-level disinfection. Noncritical items are items that contact intact skin. Noncritical items generally require low-level or intermediate-level disinfection and contact times of 1 to 10 minutes.[9]

All personnel should be educated regarding the standard hospital germicides used for disinfection and antisepsis, how they should be diluted and applied, and the hazards associated with their use. Gloves and eye protection should be worn when disinfectant solutions are handled or mixed. In large hospitals, posters on hospital walls can be used to guide selection of appropriate germicides for different situations. Disinfectant solution should be readily accessible throughout the hospital. Because mops can spread infection, cotton mops should be laundered daily, or mops with detachable microfiber heads should be used. Microfiber heads absorb a large amount of water and are not returned to the mop bucket after use. They can be laundered and reused the following day.

Methods of Sterilization

Steam sterilization. Steam sterilization involves the use of saturated steam under pressure in an autoclave to achieve sterilization. This is the most effective form of sterilization, is nontoxic and inexpensive, and as a result is the most widely used as well. The use of steam under pressure allows lower temperatures to be used for shorter periods of time when compared with dry heat sterilization (Table 11-4). The most common temperatures used for steam sterilization are 121°C (250°F) and 132°C (270°F). Cycle times vary depending on the autoclave used and whether items are wrapped or unwrapped, but are generally less than 30 minutes (see Table 11-4). There are two basic types of autoclaves: gravity displacement autoclaves, and high-speed prevacuum sterilizers. Gravity displacement autoclaves admit steam at the top or sides of the autoclave, which displaces air through a drain vent at the bottom of the chamber (Figure 11-1). High-speed prevacuum sterilizers rapidly pump air out of the sterilizer before steam is admitted. This leads to rapid penetration of steam into all surfaces. As a result, cycle times can be reduced to less than 15 minutes. Drying times are also reduced with prevacuum sterilizers.

Sterilizers should be located away from potential sources of contamination, such as sinks, trash disposal, or high-traffic areas. Before steam sterilization is performed, instruments should be cleaned thoroughly to remove organic and inorganic material, and then dried. All jointed items should be opened or unlocked, and items should not be crowded in the autoclave, so that steam can circulate freely. At least 3 inches should be left between the autoclave wall and items to be sterilized. The manufacturer's instructions for autoclave operation should be followed.

The efficacy of autoclaving must be tested for every autoclaved item, with additional quality control performed on a periodic basis. Autoclave indicator tape is routinely used; although this only indicates conditions on the outside of the package. Steam indicator strips should be included in each surgical pack and evaluated by the person opening the pack. Biological indicators provide a more definitive assessment of autoclave efficacy and should be used periodically (e.g., weekly) and the results documented. These consist of a standardized population of bacterial spores, usually on a filter paper strip or contained within a vial. The strip is then sent to the microbiology laboratory for culture, or cultured in-house, to ensure that the spores have been completely inactivated by the sterilization process. Any indicator failure should result in immediate inspection of the autoclave. Sterilization indicators should never be used as a substitute for proper autoclave operation and careful preparation, packing, and loading of equipment to be sterilized.

After steam sterilization, instruments should be allowed to dry before they are removed, which typically takes an additional 30 minutes. Items should then be stored in a location and manner that prevents further contamination.

Flash sterilization. Flash sterilization refers to the rapid sterilization of unwrapped instruments and is usually performed as an emergency procedure in an operating room setting when time is insufficient to perform the preferred sterilization of wrapped items. In general it is performed for 3 minutes at 270°F and 27 to 28 lb/in^2. Each instrument must be carefully protected to ensure it does not become recontaminated during transport back to the operating room, usually in a "flash pan." Flash sterilization has occasionally been associated with intraoperative infections and should not be used for routine disinfection purposes.

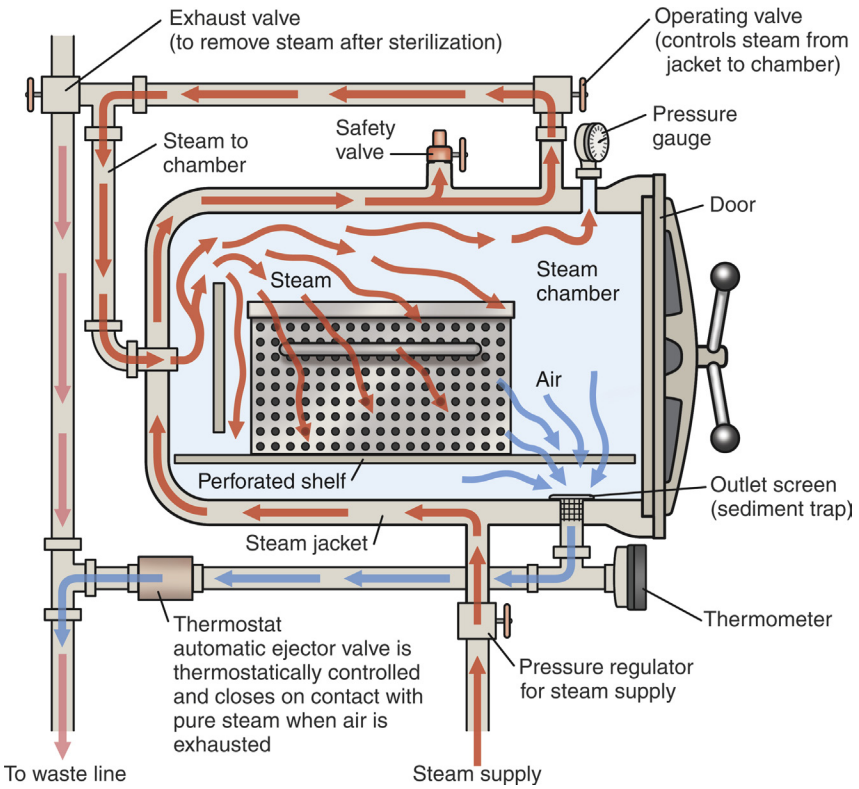

FIGURE 11-1 Gravity displacement sterilization. Steam is admitted at the top or sides of the autoclave, which displaces air through a drain vent at the bottom of the chamber.

Gas sterilization. Gas sterilization is used for sterilization of heat- and moisture-sensitive instruments but can have significant toxicity. Gases that can be used for sterilization include formaldehyde, ethylene oxide (ETO), hydrogen peroxide vapor, and ozone gases. The most commonly used gas in veterinary medicine, ETO, has strong alkylating properties and causes protein coagulation, enzyme inactivation, and damage to nucleic acid. When compared with heat sterilization, ETO sterilization takes longer (24 hours or more including the time required to allow ETO to diffuse out of packages at the end of sterilization) and is more expensive. In addition, it only achieves surface sterilization and requires sophisticated equipment and trained staff. The gas is extremely flammable, irritates the eyes and mucous membranes, is mutagenic and carcinogenic, and has a misleadingly pleasant smell.

Irradiation. Gamma irradiation uses a cobalt-60 radiation source to destroy microorganisms through generation of high-energy photons. Health care product manufacturers use gamma irradiation to sterilize disposable medical supplies, such as catheters, gloves, syringes, and pharmaceuticals.

Chemical sterilants. Chemical sterilants are used when heat or ETO gas sterilization is not available or would otherwise damage instruments, such as endoscopes or laparoscopes. Disinfectants that act as sterilants when they are used at high concentrations and for adequate contact periods include certain solutions that contain glutaraldehyde (e.g., >2.4% glutaraldehyde solutions, 0.95% glutaraldehyde with 1.64% phenol/phenate), 0.55% *ortho*-phthalaldehyde (OPA), 7.5% hydrogen peroxide, or greater than or equal to 0.2% peracetic acid (see section on Disinfectants following). Chemical sterilants must be rinsed repeatedly with sterile water and dried once sterilization is complete. Chemical sterilants must be used with the proper contact times and at the right concentration. Solutions may lose efficacy, become diluted, or become contaminated with bacteria over time. They should be replaced frequently according to the manufacturer's recommendations, and they should never be used in procedures that involve sterile body sites unless there is no other sterilizing option.

Disinfectants

Glutaraldehyde. Glutaraldehyde is a saturated dialdehyde. Glutaraldehyde solutions are relatively inexpensive, are noncorrosive, and can be used to disinfect rubber, plastics, and endoscopic equipment. Aqueous solutions require activation by alkalinization to a pH of 7.5 to 8.5 for sporicidal activity to occur. When alkalinized glutaraldehyde is used at a concentration of 2.4% for adequate periods of time, either chemical sterilization or high-level disinfection occurs, depending on the contact time (e.g., Cidex). Contact times of at least 20 minutes (at or above 20°C) are effective for high-level disinfection. Sterilization requires a 10-hour contact time and higher concentrations of glutaraldehyde (e.g., Cidex Plus, which contains 3.4% glutaraldehyde), or formulations that combine glutaraldehyde with another disinfectant.

Once activated, 2.4% glutaraldehyde solution retains activity for 14 days, provided inadvertent dilution does not occur. The solution is active in the presence of 2% organic matter. Inadvertent dilution can occur when endoscopes that contain fluid within their channels are immersed in the solution. Test strips are available from the manufacturer to monitor the activity of the solution, but should not be used to extend the solution's expiration date. They indicate inactivity when the concentration drops below 1.5%, the minimum concentration required for activity.

Glutaraldehyde irritates mucous membranes of the respiratory and gastrointestinal tracts, and so endoscopes must be rinsed properly after disinfection. It can also cause allergic contact dermatitis, but it is not mutagenic or carcinogenic. The wearing of nitrile rubber or butyl rubber gloves is recommended. Because of its relative expense and toxicity, it is not used to disinfect noncritical surfaces.

***ortho*-Phthalaldehyde.** OPA (e.g., Cidex OPA) is favored over glutaraldehyde for high-level disinfection in the United States, because it does not require activation, is stable over a wide pH range, does not cause irritation of mucous membranes, and it has a barely perceptible odor. Its activity is greater than that of glutaraldehyde, and high-level disinfection is achieved with a contact time of 12 minutes at or above 20°C.[10,11] The primary disadvantage of OPA is that it stains tissues and mucous membranes gray. Protective equipment must be worn when handling the solution, and it must be rinsed thoroughly from items after treatment. Irritation can occur with eye contact. OPA is also more expensive than glutaraldehyde.[12] Solutions can be reused for a maximum of 14 days.

Hydrogen peroxide. Hydrogen peroxide (H_2O_2) is a potent oxidizer. Hydrogen peroxide solutions are widely available, are inexpensive, and can enhance removal of organic matter. When used at concentrations of 7.5% for contact periods of at least 6 hours, hydrogen peroxide is a chemical sterilant (e.g., Sporox). High-level disinfection can be achieved with contact times of 12 to 30 minutes. Unfortunately 7.5% hydrogen peroxide causes discoloration and functional changes within endoscopes and so is not suitable for endoscope reprocessing. Although nonirritating to mucous membranes, serious ocular damage can occur with eye contact.

Accelerated hydrogen peroxide (AHP) is a patented hydrogen peroxide solution that contains surfactants, an acid, and hydrogen peroxide. Use of a 4.5% AHP gel with contact times of 10 minutes can inactivate bacterial spores.[13] Even 0.5% solutions have some sporicidal activity and can inactivate non-enveloped viruses such as canine parvovirus.[14,15] AHP has gained popularity as a disinfectant among health care institutions because it is odorless, does not generate volatile gas, is nonirritating, and is noncorrosive at dilutions used in health care settings.

Peracetic acid. Peracetic acid belongs to the peroxygen family of compounds. When used at 50°C to 56°C in a specific peracetic acid reprocessing system (Steris System 1, Steris), 0.2% solutions achieve sterilization in very short time periods (30 to 45 minutes). Peracetic acid is active in the presence of organic matter and may actually enhance its removal. Nevertheless endoscopes must still be thoroughly cleaned before sterilization to avoid fixation of blood onto the instrument. After sterilization, the processor rinses the instrument thoroughly. Peracetic acid is stable but can be corrosive and causes discoloration of endoscopes over time. It is more expensive than other chemical sterilants. Peracetic acid concentrates can cause irritation to mucous membranes and are corrosive to the eye and skin, but 0.2% solutions are generally nonirritating.

Potassium peroxymonosulfate (Trifectant, Virkon S). Like peracetic acid, potassium peroxymonosulfate is an oxidizing agent, and 1% solutions are high-level disinfectants that are capable of inactivation of non-enveloped viruses when contact times of 10 minutes are used.[16] Thus it is suitable for inactivation of canine parvovirus and feline calicivirus. Potassium peroxymonosulfate retains some activity in the presence of organic matter. Solutions are prepared from powder and remain active for 7 days. The powder is corrosive and can cause serious skin and ocular burns, but the solution is nonirritating and less corrosive than bleach. The solution stains fabric and may damage surfaces, particularly metal, over time if rinsing is not performed.

Sodium Hypochlorite (Bleach). Household bleach, which contains 5.25% to 6.15% sodium hypochlorite, is widely available, inexpensive, and active in the presence of hard water. When used at a 1:10 dilution for a 10-minute contact time, household bleach is sporicidal but is irritating and can be highly corrosive to metal surfaces. This 1:10 dilution is used to control outbreaks of clostridial diarrhea. For most hospital situations, 1:30 to 1:50 dilutions of household bleach provide more than 1000 ppm available chlorine and are effective for intermediate-level disinfection. Noncritical surfaces can be disinfected with 1:500 dilutions of household bleach (>100 ppm available chlorine), with contact times of at least 1 minute. Sodium hypochlorite is inactivated by organic matter, so cleaning is required before disinfection is performed. Solutions lose 50% of their activity over a 1-month period unless they are stored in closed brown bottles. Other disadvantages of bleach solutions are bleaching of colored fabric and the release of toxic chlorine gas when they are combined with other disinfectants such as quaternary ammonium compounds (QUATs).

Quaternary Ammonium Compounds. QUATs are often used as low-level disinfectants for noncritical surfaces in health care facilities. Although they are generally fungicidal, bactericidal, and virucidal for enveloped viruses, some bacteria resist and even grow within QUATs, and such contamination has resulted in HAIs.[17] Contact times vary by product so manufacturer recommendations should be followed; however, 10-minute contact times are often used. Advantages of QUATs include their low cost, high stability, and low toxicity. QUATs are inactivated by hard water, organic materials, soaps, and detergents.

Phenolics. Phenolics are one of the oldest known disinfectant classes. Phenol derivatives that are available for hospital disinfection include *ortho*-phenylphenol and *ortho*-benzyl-*para*-chlorophenol (e.g., Qualitrol, Vetnex). Effective contact times vary with the product. They are active against enveloped viruses and bacteria, but they are not sporicidal and have limited activity against fungi and non-enveloped viruses. Phenol derivatives are active in the presence of organic matter and hard water, are stable, and are noncorrosive. They irritate skin and mucous membranes, and have the potential to be highly toxic if ingested by cats. Because of these limitations and the availability of other effective disinfectants, phenolics are rarely used in veterinary hospitals.

Antiseptics

Alcohol. Ethyl alcohol and isopropyl alcohol are bactericidal, virucidal (for enveloped viruses), and variably fungicidal. The optimum bactericidal concentration is 60% to 90% in water (volume/volume). They do not destroy bacterial spores or penetrate proteinaceous material and so are not recommended for high-level disinfection. They can be used to disinfect noncritical items such as hospital fomites and have very low toxicity, so they are often included in waterless hand sanitizer products together with emollients to prevent drying of the skin. They are flammable and dry quickly, so it can be difficult to achieve adequate contact times (≥1 minute). As a result, alcohol should not be used for routine environmental disinfection.

Iodophors. Iodine solutions are primarily used as skin antiseptics. Formulations for disinfection are also available, which contain higher concentrations of free iodine than antiseptic

preparations. An iodophor is a combination of iodine and a solubilizing agent (e.g., polyvinylpyrrolidone in povidone-iodine), which serves to provide a sustained-release form of iodine. Dilutions of iodophors are more active against microbes than concentrated povidone-iodine, so iodophores must be diluted correctly. Iodophors are bactericidal and virucidal. Their antibacterial activity does not persist for long periods on skin or in tissues, so frequent reapplication is required. Iodophors are relatively nontoxic and nonirritating. They are stable in solution but are inactivated by organic material and they can stain plastics and, to some extent, tissues.

Chlorhexidine. Chlorhexidine is a cationic bisbiguanide that disrupts microbial cell membranes and precipitates cell contents. It is used widely for skin antisepsis in veterinary medicine. Chlorhexidine has persistent activity on the skin, is nonirritating, is active in the presence of body fluids, and has rapid bactericidal activity. Like iodophors, chlorhexidine has limited activity against fungi and mycobacteria. A 2% chlorhexidine solution is preferred over 70% alcohol or povidone-iodine for skin preparation of central venous catheter sites in human patients[18] and is the skin antiseptic of choice for collection of blood for blood cultures. Chlorhexidine may have slightly lower activity against gram-negative bacteria and fungi than povidone-iodine, and both chlorhexidine and iodophors have a slower antimicrobial activity than 60% to 90% alcohol solutions.

Principles of Cleaning and Disinfection

Standard operating procedures that describe preparation and application of disinfectants for all surfaces and objects used in a veterinary hospital, as well as waste management, should be developed, and staff and visitors should receive education about the location of protocols and their use. Wards and procedure and examination rooms should remain uncluttered in order to facilitate effective cleaning. Because organic matter can inactivate disinfectants, all visible debris should be removed before disinfection. Disinfectants are only active when applied to clean, nonporous surfaces. Porous surfaces such as dirt and wood cannot be effectively disinfected using routine procedures. Hard porous surfaces should be scrubbed with disinfectant using brushes, and then rinsed with water after the contact time has elapsed.

When hospital surfaces are cleaned, attention should be paid to corners, under cabinets, chairs, the bases of examination tables, door handles, elevator buttons, shelves, sinks, faucets, and other surfaces that might otherwise be ignored. Personal items such as cell phones and stethoscope heads should be regularly disinfected with disinfectant wipes. Cell phones should be disinfected at least daily, and stethoscope heads wiped between patients. Stethoscope tubing should be cleaned regularly with soap and water. Disinfection of other fomites should be performed on a regular (at least daily) basis. These include digital thermometers, computer keyboards and mice, land-line telephones, calculators, microscopes, otoscopes, and blood pressure cuffs.

Cleaning staff should wear gloves when general hospital disinfection is performed. This may not be necessary for spot cleaning of areas such as examination room tables or fomites. Depending on the disinfectant, additional protective attire, such as a gown, boots, or face mask, may be required if there is a probability of significant splashing during the disinfection process. After disinfection is complete, protective clothing should be removed and handwashing performed. The individuals responsible for cleaning and disinfection at each time point, situation, and location in the hospital should be clearly identified. A plan for the cleaning of outdoor areas that become contaminated with excretions or other biohazardous material should be outlined.

When disinfection of cages or runs is performed, animals should be removed and placed in a clean holding cage or run, away from other patients. Solid waste and soiled laundry or paper should be removed and placed directly in waste or laundry containers, taking care to avoid dripping onto the floor. If laundry is contaminated with potentially infectious material, it must be bagged and labeled with the contents and suspected infectious agent. Proper contact times should be used after application of disinfectant solution. Runs and cages should be allowed to dry completely before a new patient is introduced.

Immunocompromised People and Children

People are considered immunocompromised if they (1) have various comorbidities such as diabetes mellitus, chronic kidney failure, leukopenia, immune-mediated disease, HIV/AIDS, congenital immunodeficiencies, hepatic cirrhosis, cancer, or splenectomy; (2) are being treated with immunosuppressive drugs or chemotherapeutics; (3) are very young (5 and under) or very old; or (4) are pregnant, although the immunosuppressive effects of pregnancy are considerably lower than those of the preceding disorders.

Immunocompromised staff members who work in small animal hospitals should discuss any necessary work restrictions and precautions in light of their specific condition with their physician or an infectious disease doctor. The risks for a diabetic, for example, may be significantly different from those for a transplant recipient. In general, immunosuppressed individuals who work in small animal hospitals should avoid handling patients with suspected or known infectious diseases. Gloves should always be worn when handling animals and animal fluids or excreta, and strict attention to general hand hygiene and any bites, scratches, or sharps injuries is critical. Children 5 years of age or younger should be kept out of patient care areas. Strict attention to hygiene and protective apparel is necessary when handling soiled cat litter or animal feces, because of the risk for transmission of enteropathogens. The reader is referred to other chapters in this book for public health implications of specific infectious diseases that may be encountered in small animal hospitals. If possible, small animal clinics should establish a relationship with a doctor who is prepared to consult on zoonotic exposures with staff members.

Transmissible Disease Surveillance and Reporting

A transmissible disease surveillance program allows collection of baseline data that establishes the prevalence of certain infectious diseases, so that possible outbreak situations can be readily identified. It can also provide background prevalence information on antimicrobial drug susceptibility patterns, which can assist in the initial selection of antibacterial drugs for individual animal patients while culture and susceptibility test results are in progress. Information is generally collected for multidrug-resistant bacterial infections, zoonotic diseases, highly contagious diseases, pathogens that are difficult to inactivate with

disinfectants, or agents of regulatory concern. Although surveillance may seem like a difficult, time-consuming, and expensive measure, in reality, it can be easy and cost-effective and can provide important information.

There are two main forms of infection control surveillance applicable to veterinary hospitals: active and passive. *Active surveillance* involves collection of data specifically for infection control purposes. This can provide the highest quality and most relevant information, but it can be expensive and time consuming. Examples of this would be collection of swab specimens to screen for infection with methicillin-resistant *Staphylococcus pseudintermedius* (MRSP) from dogs before surgery as part of MRSP outbreak investigation. Active surveillance is a core component of infection control in most human hospitals, but it is only sporadically used in veterinary medicine and is rarely needed in most veterinary clinics. It is typically reserved for large facilities with increased infection control risks and personnel available to direct such testing, or as a part of outbreak control.

In contrast, *passive surveillance* is a practical, easy, and cost-effective surveillance approach that can and should be performed in every veterinary clinic. It involves the use of data that are already available, such as information about surgical site infections collected during routine follow-up or culture results from clinical testing. The quality of passive surveillance data can be limited by poor or incomplete record keeping or sporadic use of appropriate diagnostic tests, but if properly collected and if potential biases are understood, passive surveillance data can provide important insight into aspects such as endemic disease rates, common pathogens, and antimicrobial susceptibility trends. To facilitate passive surveillance, clinicians should be encouraged to use appropriate diagnostic testing to determine the etiology of nosocomial infections, even if the clinical consequences are not severe. They also should be encouraged to confirm a diagnosis in animals with suspected transmissible disease. This allows clients to protect their other animals and their families and friends who might be in contact with the pet, and it benefits the hospital.

Another form of surveillance that is easy to perform and potentially very useful is *syndromic surveillance*. This involves surveillance for readily identifiable syndromes (i.e., diarrhea, cough) instead of specific diagnoses. Although syndromes do not indicate a specific disease, they can indicate an increased risk of infectious disease. Syndromic surveillance is an initial screening tool that can be used by all personnel, including lay personnel, to flag potentially high-risk cases and allow for early implementation of enhanced infection control practices. All clinic personnel should be made aware of certain syndromes that indicate the need either for isolation or for further investigation, such as diarrhea, fever of unknown origin, acute neurologic disease, wound infections, and acute respiratory tract disease. Protocols to deal with these animals on arrival should be developed. As an example, a dog with an acute onset of cough should be considered potentially infectious, and if front office personnel note this syndrome at the time the appointment is made, the dog can be handled appropriately on arrival (e.g., admitted directly to isolation or an examination room, with personnel wearing enhanced barrier precautions from the onset). The role of lay staff (i.e., front office stall) is critical, as these people are the ones who are most able to identify such cases before they enter the clinic and ensure that they are properly handled to prevent nosocomial or zoonotic transmission.

BOX 11-3

Infectious Diseases of Dogs and Cats That Are Reportable or Potentially Reportable to Public Health Agencies

Amebiasis
Granulocytic anaplasmosis
Brucellosis
Campylobacteriosis
Coccidioidomycosis
Cryptococcus gattii infection
Cryptosporidiosis
Giardiasis
Leishmaniosis
Leptospirosis
Lyme disease
Q fever
Rabies
Salmonellosis
Tularemia
Tuberculosis
MRSA (not MRSP)
Novel H1N1 influenza virus infections
West Nile Virus infection
Yersinia infections

Environmental cultures are rarely informative and so are not considered a useful routine infection control tool. They may be used to detect a specific pathogen if an outbreak of an HAI is suspected when there is suspicion that the environment is a source of exposure. However, it is often difficult to distinguish cause from effect (i.e., environmental contamination that leads to transmission vs. environmental contamination that occurs as a result of contamination from a patient in the absence of a risk of transmission).

Although surveillance programs require time, effort, and expense, in the long term they may save morbidity and mortality and reduce costs that relate to control of large outbreak situations or the legal ramifications of HAIs.

When zoonotic diseases are identified, all in-contact individuals and, if necessary, public health authorities should be notified in the appropriate manner. The specific diseases that must be reported to authorities vary among geographic locations. Examples of diseases that may be of interest to public health authorities are listed in Box 11-3.

Surgical Preparation

Surgical site infections (SSIs) are an uncommon but important and sometimes devastating complication of surgery. Every patient undergoing a surgical procedure is at some risk of SSI. Standard practices have been developed to reduce the risk of SSIs. Although a wide range of practices are relevant, preparation of the patient and preparation of the surgeon are critical.

Patient Preparation

The patient's endogenous microflora is an important source of pathogens that cause SSIs. Careful preparation of the patient, therefore, can help reduce contamination of the surgical site during surgery. The goal of preoperative surgical site management is to eliminate potential pathogens while not creating a physical environment that is more conducive to bacterial colonization or infection postoperatively. Bathing of the patient preoperatively is reasonable if there is significant contamination of the haircoat[19] and if the patient's coat can be dried by the time of surgery. In most situations, bathing is not required. Rather, careful hair removal and skin antisepsis are the main measures.

The goals of surgical scrubbing of patients are to reduce bacterial counts, reduce debris, and facilitate later antisepsis. Scrubbing should be done as atraumatically as possible. Minimizing skin damage during clipping and scrubbing is essential, because skin damage from excessive attempts to clip all remaining pieces of hair or from forceful scrubbing of the surgical site can create an environment that is more amenable to bacterial growth. This predisposes to infection rather than reducing the risk. Clipping should be performed after anesthesia, to reduce the risk of trauma associated with a struggling patient and to minimize the time between clipping and surgery. Clipping should be done outside the operating environment. There is currently no information that relates to optimal methods of cleaning and disinfecting clippers. Repeated use of clipper blades without sterilization not surprisingly results in higher levels of bacterial contamination of blades[20]; however, the clinical relevance of this is unclear, because the surgical site is cleaned and disinfected after clipping. Regular cleaning and disinfection of clippers are probably useful, and they should be thoroughly cleaned and disinfected after use on an animal with a potentially transmissible infection (e.g., an animal with diarrhea), on an area where the skin is broken, or on any area where the skin or hair is significantly contaminated with feces, urine, blood, or other body fluids.

After hair removal, various approaches for skin antisepsis can be used. There is little outcome-based evidence of the relative efficacy of different approaches in the human literature. Typically, a three-step process is used, with initial scrubbing of the site with biocidal (i.e., chlorhexidine, povidone-iodine) soap, followed by application of alcohol for biocidal effects and to remove oils, and a final application of a biocidal solution with residual activity (e.g., chlorhexidine).

Surgeon Preparation

The surgeon's body and clothing are potential sources of SSI pathogens, and standard practices have been developed to reduce the risk of contamination of the surgical site. One aspect is the use of proper protective clothing. Every person in the operating environment should wear clean surgical scrubs. These scrubs should be dedicated for use only in the operating room or should be covered with a clean laboratory coat whenever the individual is outside of the operating room.

Surgical hand antisepsis is critical because of the close contact of the hands with the surgical site and the relatively high incidence of grossly evident breaks or micro-breaks in gloves. Surgical hand antisepsis is designed to greatly reduce bacterial burdens on the hands, particularly the abundant transient microflora that contains most of the relevant pathogens that cause SSIs. Surgical antisepsis must find a balance between effective elimination of pathogens, minimization of trauma to the skin (because skin irritation facilitates bacterial growth), and time constraints. The traditional approach to surgical hand antisepsis has involved structured scrubbing of the hands for a predetermined time. Recommended scrub times vary between products but are typically 2 to 4 minutes.

Application of alcohol-chlorhexidine combinations has been evaluated as a replacement for surgical scrubbing and has been shown to be more effective than standard surgical scrub methods.[21,22] Specific manufacturer instructions should be followed, and hands and arms must be dry before application of gloves. The use of alcohol-based surgical hand antisepsis products is encouraged as a replacement for traditional scrubbing.

BOX 11-4

Example of Criteria Specified within a Hospital for Use of Restricted Antimicrobial Drugs

- Infection must be documented based on clinical abnormalities *and* culture.
- Subclinical infections should not be treated with these antibiotics. These antibiotics should be reserved for infections that are life threatening.
- Resistance to all other reasonable options and susceptibility to the chosen antimicrobial must be documented.
- The infection must be potentially treatable.
- The clinician should contact the infectious disease control officer or a clinical microbiology faculty member by email or telephone to discuss antimicrobial susceptibility test results, determine whether there are any other viable options, and confirm that a restricted antimicrobial is necessary.

Regardless of the method used, a thorough handwash with careful cleaning under the fingernails must be performed at the beginning of each day.[23] Long (>¼-inch) and artificial nails are prohibited in many human health care facilities and some veterinary hospitals because they harbor pathogenic bacteria[24] and are associated with surgical glove tears.

A cap and mask must be worn during hand antisepsis, and sterile gown and gloves must then be donned using appropriate technique.

Antimicrobial Drug Use

The use of antimicrobial drugs contributes to the selection for antimicrobial-resistant bacteria. Inclusion of a policy on antimicrobial drug use in the infection control protocol may help to reduce the prevalence of multidrug-resistant HAIs. On their own, fever and leukocytosis are not justification for antimicrobial drug treatment. Whenever possible, when infection is suspected, attempts to obtain material for culture and susceptibility testing should be made before antimicrobial drug treatment is initiated. When secondary bacterial infection is likely, attempts to identify and treat the underlying cause should be made. In an effort to minimize the emergence of resistance in bacterial flora of animals in the hospital and decrease the risk of resistant HAIs, the use of several antimicrobials (e.g., certain parenteral third-generation cephalosporins, vancomycin, linezolid, and carbapenems such as imipenem and meropenem) may be restricted unless criteria must be met (Box 11-4).

Prevention of Infectious Disease in Shelters and Breeding and Boarding Facilities

Attention to hand hygiene and proper disinfection of fomites and the environment as outlined for hospitals can also be used to reduce transmission of infectious pathogens in shelters and in breeding and boarding facilities (Box 11-5). Disinfectants with activity against parvoviruses and feline calicivirus should be used routinely, such as a 1:30 dilution of household bleach,

BOX 11-5

Factors That Should Be Considered for the Reduction of Infectious Disease Transmission in Animal Housing Facilities

- Adequate ventilation and air quality
- Adequate temperature
- Adequate lighting conditions (including provision of darkness at night)
- Population density/space per animal
- Daily removal of fecal and urine contamination and disinfection
- Adequate drainage
- Separate housing areas for dogs and cats
- Isolation for sick animals
- Separate housing areas for young and adult animals
- Individual housing or small groups of two to four compatible animals
- Separation of elimination, feeding, and resting areas
- Elevated resting areas
- Elevated cages for cats
- Provision of hiding areas
- Sound control
- Use of surfaces that can be readily disinfected
- Proper use of appropriate disinfectants
- Hand hygiene and protective clothing
- Fomite control
- Use of appropriate vaccination protocols
- Order of care (young animals, healthy adults, and then sick animals)
- Proper nutrition
- Pain management
- Free access to clean water
- Rodent and pest control
- Parasite control
- Daily monitoring by properly trained individuals
- Proper diagnosis of disease outbreaks

potassium peroxymonosulfate, or accelerated hydrogen peroxide solutions. Removal of organic matter, the use of surfaces that can be adequately disinfected, and proper contact times are essential.

Factors that increase stress should also be minimized, such as high population densities, the grouping of animals, and poor nutrition. Cats and dogs should be separated from one another. In cats, the use of methods that reduce stress, such as provision of a low-stress cage environment, can dramatically reduce the frequency of upper respiratory tract disease. Compartmentalized housing allows animals to urinate and defecate away from areas where they rest and eat, and provision of a hiding area so that animals can retreat from visual stimulation can also reduce stress. A total of 10 to 20 air changes per hour is often recommended for animal housing, but higher levels of ventilation may be needed with increased population density and concentration of airborne contaminants. Cages should be cleaned and disinfected at least daily. The use of compartmentalized housing areas also facilitates cleaning and disinfection without requiring an animal to be removed from the housing area. Isolation areas for sick animals should be present and these should have separate airflow from areas that house healthy animals. Mass treatment of dogs and cats with upper respiratory tract disease with antimicrobials without attention to the underlying causes only results in selection for antimicrobial drug resistant organisms and is not recommended. Detailed information on the control of infectious diseases in shelter environments is beyond the scope of this book but can be found elsewhere.[25] The American Association of Shelter Veterinarians and the Humane Society of the United States have published guidelines for operation of animal shelters.[26,27]

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