Infection Prevention and Control Best Practices

For Small Animal Veterinary Clinics

Dear veterinary staff member,

We are conducting a brief online survey to understand your current infection control practices and your motivation for seeking out these guidelines. Participation in the survey is strictly voluntary. You may exit the survey at any time, and you may skip any questions you wish. All responses are anonymous

This survey should only take approximately 2-3 minutes to complete. We would greatly appreciate your feedback.

Click this link to start the survey:

https://uoguelph.eu.qualtrics.com/jfe/form/SV_6DbIPOk3dZnLbQ9

ONTARIO ANIMAL HEALTH NETWORK

Infection Prevention and Control Best Practices

For Small Animal Veterinary Clinics

CANADIAN VETERINARY MEDICAL ASSOCIATION
L'ASSOCIATION CANADIENNE DES MÉDECINS VÉTÉRINAIRES

One Health at Guelph.
CPHAZ
Centre for Public Health and Zoonoses

January 2020

ONTARIO ANIMAL HEALTH NETWORK

One Health at Guelph.
CPHAZ
Centre for Public Health and Zoonoses

Infection Prevention and Control Best Practices

For Small Animal Veterinary Clinics

M.E.C. Anderson, M. Wimmers, J.S. Weese

The authors would like to acknowledge and express appreciation to the contributors to the 1st edition of these guidelines, as well as the following groups and individuals whose input was considered in the 2nd edition:

- The Canadian Veterinary Medical Association, National Issues Committee (Dr. Shane Renwick) for endorsement, and for allowing distribution through its website.
- Dr. Brandy Burgess
- Dr. Devon Metcalf
- Dr. Lisbeth Rem Jessen
- Dr. Jason Stull
- Dr. Ulrika Grönlund

On behalf of the Canadian Veterinary Medical Association:

- Dr. Nigel Gumley
- Dr. Sherlyn Spooner

Disclaimer

This best practices document is intended to guide clinical practice only and provide assistance for decision-making on infection prevention and control issues. Its use should be flexible to accommodate specific challenges and risks in different facilities and regions while helping to ensure best practices in infection prevention and control. These practices neither constitute a liability nor discharge from liability. While every effort has been made to ensure accuracy of the contents at the time of publication, the authors do not give any guarantee as to the accuracy of information contained herein, nor accept any liability, with respect to loss, damage, injury or expense, arising from any errors or omission in the contents of this work.

Copyright

Anderson MEC, Wimmers M, Weese JS. Infection Prevention and Control Best Practices for Small Animal Veterinary Clinics, 2nd ed. Guelph: Ontario Animal Health Network, 2019.

First edition, November 2008.

Centre for Public Health and Zoonoses

There is an urgent need for public health research at the human-animal-environmental interface, and for interdisciplinary research approaches spanning from basic laboratory sciences to applied field studies, and from animals to humans. The Centre for Public Health and Zoonoses (CPHAZ) at the University of Guelph provides focus and leadership for research, education, and knowledge dissemination in public health at the human-animal-environment interface. Collaborations of CPHAZ members at the University of Guelph and the Ontario Veterinary College with external collaborators and partners is a key function of the operation of CPHAZ. Through their activities, members of CPHAZ help to identify and solve problems and implement solutions in public health at the human-animal-environmental interface, contribute to rapid response to new and emerging zoonotic diseases, and highlight the societal relevance of veterinary medicine in public health.

For more information, visit www.cphaz.ca.

Ontario Animal Health Network

The Ontario Animal Health Network (OAHN) was created to achieve coordinated preparedness, early detection, and response to animal disease in Ontario, through sustainable cross-sector networks. OAHN is a “network of networks” with individual networks for different species/sectors, each of which involves collaboration among veterinarians, animal owners and other stakeholders in the field with laboratory, academic and government experts.

OAHN gathers information from each sector and available laboratory data throughout the year and sends out reports to veterinarians and other stakeholders as appropriate for each sector, highlighting trends and current animal health topics (particularly related to infectious disease) and helpful resources pertinent to each species. The goal is help veterinarians stay abreast of emerging issues, and to provide resources to help educate animal owners and clinic staff about infection prevention and control.

For more information visit www.oahn.ca.

Prepared by

Maureen E.C. Anderson DVM, DVSc, PhD
Madison Wimmers BSc, MPH
Ontario Animal Health Network
Guelph, Ontario

J. Scott Weese DVM, DVSc
Centre for Public Health and Zoonoses
Department of Pathobiology, University of Guelph
Guelph, Ontario

Summary of updates and additions to 2nd edition

- Updates throughout all chapters

- New sections within existing chapters include:
  - Surveillance: Syndromic surveillance
  - Hand Hygiene: Compliance
  - Patient Care and Handling: High risk admissions
  - Cleaning and Disinfection: Sterilization
  - Safety of Clinic Personnel: Clinic laboratory, high risk personnel
  - Dental Procedures: Equipment disinfection, antimicrobial prophylaxis
  - Non-Patient Animals: Boarders/day care, staff pets, blood donor animals, research/teaching animals

- New chapters that have been added include:
  - Antimicrobial Stewardship
  - Hospital Associated Infections and Other Infectious Syndromes
  - Blood Donation
  - Rehabilitation and Physical Therapy

Table of Contents

Section 1: Introduction 7

- Basic Principles of Infection Prevention and Control 8
  - General concepts 8
  - Rationale for routine practices – The chain of transmission 9
  - Hierarchy of infection control measures 12
- The Infection Prevention and Control Program 13
- Surveillance 14
  - Passive surveillance 15
  - Active surveillance 15
  - Syndromic surveillance 15
- Antimicrobial Stewardship 17

Section 2: Routine Practices 19

- Hand Hygiene 19
  - When to perform hand hygiene 19
  - Hand washing with soap and water 19
  - Alcohol-based hand sanitizers 20
  - Skin care 21
  - Compliance 21
- Personal Protective Equipment 23
  - Personal protective outerwear 23
  - Additional personal protective equipment 25
- Patient Care and Handling 29
  - Isolation facilities 29
  - Wound care and bandages 31
  - Feeding of raw meat diets and treats 31
  - High risk admissions 32
- Hospital-Associated Infections and Other Infectious Syndromes 34
  - Bloodstream infections 35
  - Catheter-associated urinary tract infections 35
  - Other sources of HAIs 36
  - Management of specific infectious syndromes 37
- Cleaning, Disinfection, Sterilization 41
  - Cleaning 41
  - Disinfection 42
  - Sterilization 51
- Laundry and Waste Management 56
  - Laundry management 56
  - Waste management 58

Section 3: Special Procedures 60

- Surgery 60
  - Surgical environment and suite design 60
  - Personnel considerations 61
  - Equipment considerations 64
  - Perioperative antimicrobials 65
  - Surgical site management 67
  - Surgical site infection surveillance 68
- Dental Procedures 72
- Blood Donation 73
- Rehabilitation and Physical Therapy 74
  - Patient assessment 74
  - Hydrotherapy 74
  - Dry rehabilitation therapy 77

Section 4: Additional Considerations 78

- Safety of Clinic Personnel 78
  - Bites and scratches 78
  - Sharps 79
  - Clinic laboratory 80
  - Necropsies 82
  - Vaccination of personnel 83
  - High-risk personnel 83
  - Training and education of personnel 84
- Client Visitation 85
- Non-Patient Animals 86
  - Boarding / day care 86
  - Staff pets 87
  - Clinic pets 87
  - Blood donor animals and colonies 88
  - Research and teaching animals 88
- Education 89
- Reportable Diseases 91
- Clinic Design 92
- Vector Control 94
  - Ticks 94
  - Other ectoparasites 95

Appendices

- Management of Rabies Suspects 96
- Surgical Safety Checklist 98
- Clinic IPC Audit Checklist 99

Table of Contents continued

Tables, Figures, Boxes

Section 1

- Figure 1. How microorganisms are transmitted 11
- Box: Considerations for clinic infection surveillance programs 14
- Box: Information to assess when booking appointments for sick patients 16
- Box: Possible components of a clinic antimicrobial stewardship program 17

Section 2

- Figure 2. How to remove a gown 24
- Box: Procedure for doffing of personal protective equipment 24
- Box: Precautions to help reduce the risk of a bloodstream infection when placing a catheter 35
- Table 1. Recommended personal protective equipment for routine veterinary procedures 26
- Table 2. Infectious disease control precautions by disease condition and agent 27
- Table 3. Recommended cleaning and disinfection procedures for common environmental surfaces 43
- Table 4. Spaulding's classification of medical equipment / devices and required levels of processing and reprocessing 45
- Table 5. Common chemical disinfectants 45
- Table 6. Characteristics of selected disinfectants 49
- Table 7. Antimicrobial spectrum of selected disinfectants 50
- Table 8. Sterilization methods 52

Section 3

- Table 1. Surgical wound classifications 65
- Table 2. Commonly used perioperative antimicrobials for small animal surgical procedures 66
- Table 3. Definitions of different categories of surgical site infections 69
- Table 4. Examples of surgical site infection (SSI) surveillance methods 70
- Table 5. Criteria for identification of veterinary patients that may require antimicrobial prophylaxis prior to dental procedures 72

Section 4

- Table 1. Laboratory biosafety resources 82
- Table 2. Important considerations when determining whether boarding should be permitted at a veterinary facility 87

Introduction

Veterinary facilities face many of the same infection prevention and control challenges that human healthcare facilities encounter. Hospital-associated infections (HAIs) can have devastating effects on the health of veterinary patients, as well as the emotional and financial well-being of their owners. Outbreaks of HAIs can have a significant impact on patients, their owners and veterinary personnel. Additionally, the close contact between most people and their pets allows for transmission of infectious agents between humans and animals, in both directions, and many of the most important HAIs in human hospitals are now emerging in veterinary hospitals. Veterinary clinics can act as reservoirs of human and animal pathogens and play a role in dissemination of infectious agents including antimicrobial-resistant bacteria into the general population, with potential effects on human and animal health. Veterinary personnel also face an inherent risk of zoonotic diseases from contact with both healthy and ill animals. All these issues clearly indicate why infection prevention and control is an important part of veterinary practice. However, the field of veterinary infection control is still relatively underdeveloped compared to that of infection control in human healthcare, and limited resources are currently available to help veterinarians design and implement adequate infection control programs.

Purpose

The purpose of this document is to provide veterinary personnel with a succinct guide to principles and practices of infection control relevant to small animal veterinary clinics. This document provides the basic information needed to develop an infection control program and establish basic infection control practices for such a clinic, with specific emphasis on critical routine practices such as hand hygiene, and cleaning and disinfection.

Scope of document

This document pertains to infection prevention and control in small (companion) animal veterinary clinics and is relevant to all personnel who work in association with such clinics, including veterinarians, veterinary technicians and lay staff. For the purposes of this document, "veterinary personnel" refers to all personnel who work in a veterinary clinic. This includes non-clinical staff, as in many situations these individuals may still have periodic direct or indirect contact with patients and pathogens within a clinic.

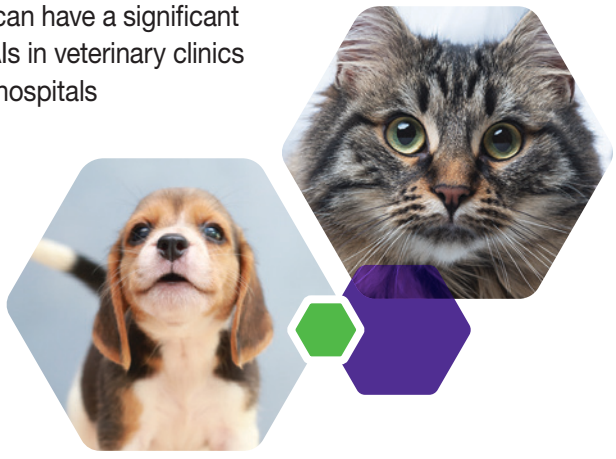
Guiding principles

1. Infection prevention and control strategies are designed to protect patients, owners, veterinary personnel and the community.

2. A significant percentage of hospital-associated infections (HAIs) in veterinary clinics can likely be prevented with proper compliance to basic, practical infection control practices.
   - Although poorly quantified, HAIs occur in veterinary clinics and can have a significant impact on animal health. While the proportion of preventable HAIs in veterinary clinics is unknown, it has been estimated at 30-70% of HAIs in human hospitals are preventable (Haley 1985).

3. A systematic approach to infection prevention and control requires all veterinary personnel to play an active role in protecting every person and animal associated with the veterinary clinic, patients or veterinary personnel.

4. Veterinary personnel need to follow infection prevention and control protocols at all times and use critical thinking and problem solving in managing clinical situations.



Basic Principles of Infection Prevention and Control

General concepts

Every veterinary clinic, regardless of size and type, should have a documented infection control program. This may range from a simple written collection of basic infection control practices, to a formal infection control manual with specific training, monitoring, surveillance and compliance programs. Lack of a clearly defined infection control program may lead to unnecessary patient morbidity and mortality, and exposure of veterinarians, staff and owners to zoonotic pathogens. Improved infection control is a necessity as veterinary medicine evolves. Advances in veterinary medicine mean that animals are living longer, and owners are often expecting a higher level of care for their pets that is more comparable to what they themselves may receive. There are also more animals at higher risk for infection in general because of more invasive and immunosuppressive therapies. In addition to the desire to achieve "best practice" standards whenever possible, the increasingly litigious nature of society may be one of the driving forces toward improved infection control in veterinary clinics. While the potential liability associated with morbidity and mortality in individual pets is limited, the potential consequences of zoonotic diseases in owners and staff are significant and warrant careful consideration.

Infection prevention and control measures can be broadly divided into three main categories: those that decrease host exposure, decrease host susceptibility and increase host resistance to infectious pathogens.

1. Decreasing **exposure** is the most important aspect of disease control in most situations. If a pathogen does not encounter an individual, then disease cannot occur. The number of organisms to which a host is exposed is also an important factor in determining whether or not colonization or infection (disease) will ensue. Depending on the pathogen, decreasing or preventing exposure may be easy, difficult or impossible.

2. There are many factors that interact to determine whether or not infectious disease will develop in a particular host. In most cases, simple exposure of an animal to an infectious agent does not mean that disease will result. The **susceptibility** of the individual to a particular number of an infectious agent plays an important role. Although difficult to quantify, certain situations may result in increased susceptibility to infection and disease. Many factors causing increased susceptibility are not preventable, but some are, and efforts should be undertaken to address these issues. Factors to consider include judicious use of antimicrobials and other drugs, provision of proper nutrition, adequate pain control, and appropriate management of underlying disease.

3. Measures to actively increase **resistance** of a host are commonly used in veterinary medicine, but these should be considered only the third line of defense, after those meant to decrease exposure and susceptibility. Vaccination is currently the main technique used to increase resistance of animals or humans to infection. However, no vaccine is 100% effective. Therefore, while vaccination is an important part of infection prevention and control, it must not be the only component of an infection control program if the program is to be successful. In addition, many HAI-infections are caused by opportunist microorganisms for which vaccines are unavailable.

Rationale for routine practices — The chain of transmission

Transmission of infection during the provision of health care requires three elements: a source of infectious microorganisms, a **susceptible host**, and a **means of transmission** for the microorganism. Prevention of infection in animal health care settings should be directed primarily at interrupting the transmission of microorganisms from source to host, because agent and host factors are typically more difficult to control.

Source

Animal sources of infectious microorganisms may be animals which are merely colonized by an infectious agent (meaning the pathogen resides in or on the body, but is not associated with any clinical disease or host response), animals in the pre-clinical (incubation) phase of disease, animals with acute disease, animals with chronic disease caused by persistent infection, and animals that are recovering from clinical disease but are still shedding the infectious agent. People can be an important source of zoonotic pathogens, and like animals they may be colonized or infected. Contamination on a person's clothing or body, particularly the hands, can also be a source of infectious microorganisms. Other potential sources include food, water, and an animal's own indigenous microflora, which may be difficult to control. Inanimate objects, including medical equipment, supplies and drugs, animal bedding, environmental surfaces and waste that have been contaminated can also be important sources. Microorganisms to consider include bacteria, viruses, fungi and parasites. In some cases, vectors such as lice, mosquitoes, flies, ticks, fleas, rodents and other vermin can transmit certain pathogens.

Host

Decreasing host susceptibility:

Decreasing host susceptibility to infection is difficult to achieve in a hospital setting. Regarding patients, the judicious use of antimicrobials, minimizing the use of immunosuppressive agents, avoidance of dietary changes whenever possible, ensuring adequate nutritional intake, adequate pain control, and limiting the use of invasive devices should be considered, as these can all have an impact on host immune function. For hospital personnel, it may not be possible to directly decrease their own susceptibility to infection, but it is important to be aware of those individuals who may have increased susceptibility. These include persons who are immunosuppressed due to disease or medical treatment, or who are being treated with antimicrobial drugs, have open wounds or who are pregnant. Good communication between veterinary personnel, their physicians and clinic administration is important to lessen the risk of zoonotic infection.

Increasing host resistance:

Vaccination is currently the main technique used to increase resistance of animals and humans to infection. As noted, no vaccine is 100% effective and there are many pathogens for which vaccines are unavailable. Factors to consider when developing vaccination recommendations or requirements include the prevalence of a particular disease in the area, risk to healthy and compromised patients, transmissibility of the disease, risk to veterinary personnel, ability to treat the disease, efficacy of vaccination and safety of vaccination. Vaccination can only be maximally effective when it is used in conjunction with other appropriate infection control practices.

Transmission

Microorganisms are transmitted in animal health care settings by four main routes: contact, droplet, air-borne and vector-borne transmission. The same microorganism may be transmitted by more than one route.

1. **Contact transmission** is the most important and frequent mode of transmission of hospital-associated infections (HAIs). It can be divided into direct and indirect contact transmission.
   - **Direct contact transmission** involves direct body surface-to-body surface contact resulting in physical transfer of microorganisms from an infected or colonized animal. For example, two dogs in a waiting room that come into direct contact when they sniff each other may transmit pathogens present in their noses or perineal areas; direct contact of a veterinarian's hands with a wound on an animal may result in transmission of opportunistic pathogens from the normal microflora of the person's hands, or infectious organisms present in the animal's wound, to the patient or the veterinarian, respectively.
   - **Indirect contact transmission** is the result of physical transfer of microorganisms from the original animal (or human) source to a new host, without direct contact between the two. This typically involves body surface contact with an inanimate object, environmental surface or the integument of another animal or person that has been transiently contaminated by the original animal (or human) source. For example, handling one animal and then petting another animal without washing one's hands constitutes indirect contact between the two animals.

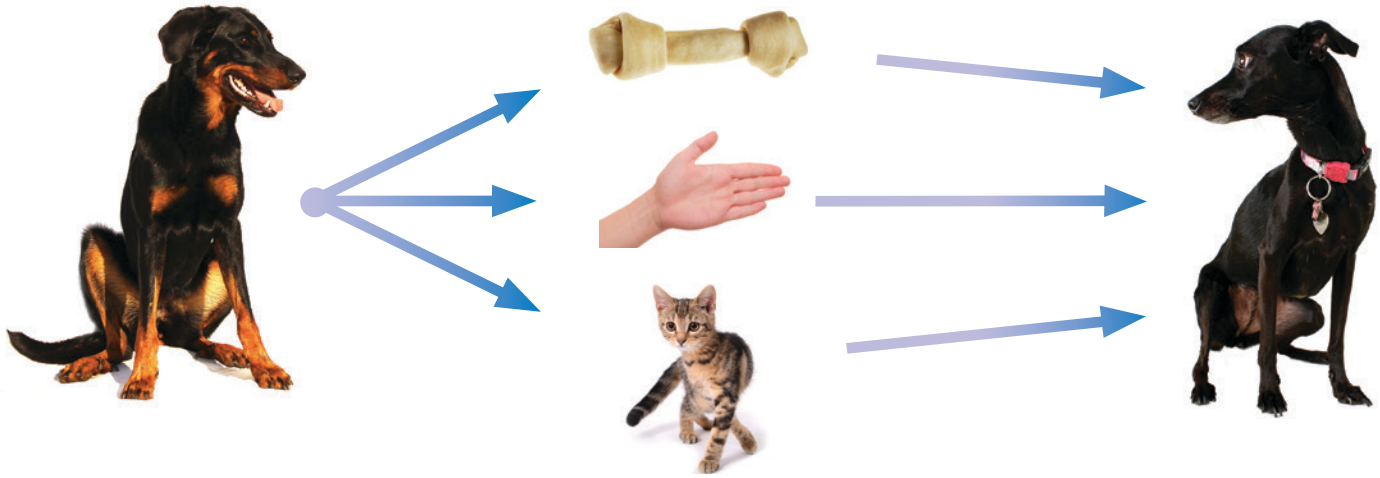
2. **Droplet transmission** is theoretically a form of contact transmission. However, the mechanism of transfer of the pathogen from host to host is quite distinct from either direct or indirect contact transmission. Droplets are generated from the source animal primarily during coughing or sneezing, and during the performance of certain procedures such as suctioning. Transmission occurs when droplets containing microorganisms generated from the source animal are propelled a short distance through the air (usually less than one meter) and deposited on the new host's conjunctiva (i.e. in the eye), nasal mucosa, mouth, or an open wound. For example, a cat with an upper respiratory tract infection can transmit viruses or bacteria to another cat in the waiting room by sneezing on it, particularly if they are face-to-face, even if the animals do not touch each other directly. Because droplets do not remain suspended in the air, special air handling and ventilation are not required to prevent droplet transmission; that is, droplet transmission must not be confused with air-borne transmission. Droplets can also contaminate the surrounding environment and lead to indirect contact transmission.

3. **Airborne transmission** occurs by dissemination of either airborne droplet nuclei (5 µm or smaller, about 2-3 times the size of most bacterial pathogens) from partly-evaporated droplets containing microorganisms, or dust particles containing the infectious agent. Microorganisms carried in this manner remain suspended in the air for long periods of time and can be dispersed widely by air currents. They may be inhaled by another host within the same room, or they may reach hosts over a longer distance from the source, depending on environmental factors. Airborne transmission of pathogens in veterinary clinics is very rare.

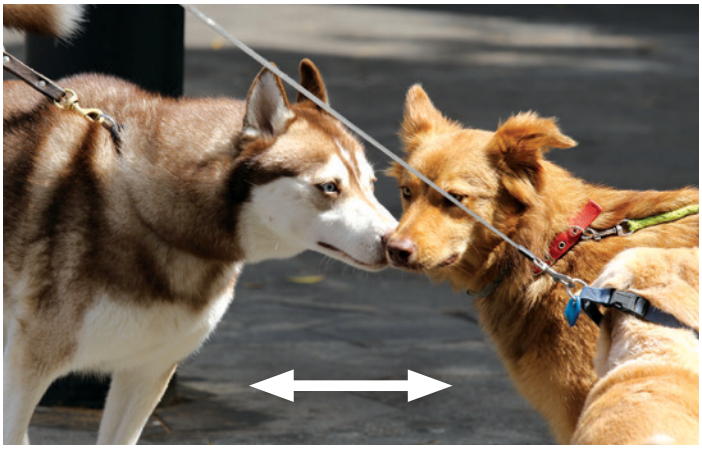
4. **Vector-borne transmission** occurs when vectors such as mosquitoes, flies, ticks, fleas, rats, and other vermin transmit microorganisms. Some act as simple mechanical vectors, comparable to indirect contact transmission, whereas others acquire and transmit microorganisms by biting. It is important to have control measures in place to reduce or eliminate the presence of such vectors in veterinary clinics.

FIGURE 1. How microorganisms are transmitted

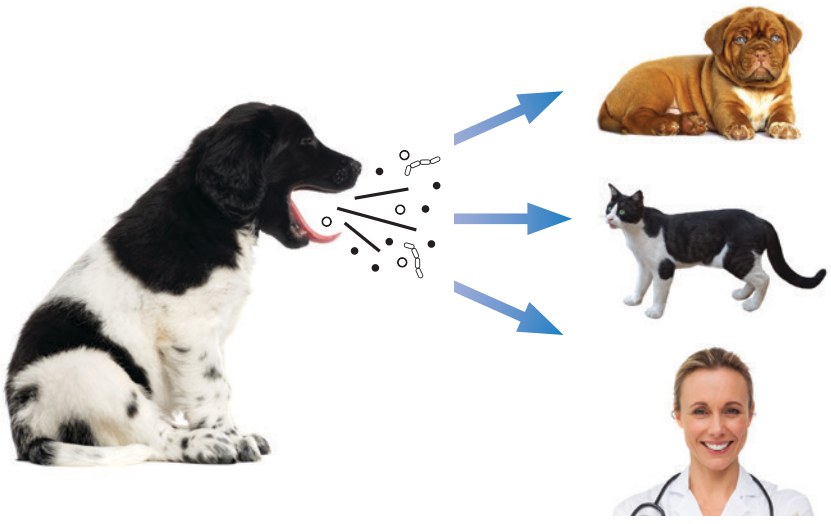
Indirect Contact



Direct Contact

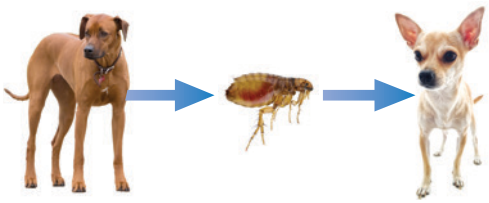


Droplet (<1 metre)



Airborne (>1 metre)

Vectorborne



Hierarchy of infection control measures

The coordinated efforts of occupational health and safety groups and building engineers have created a framework in human medicine that includes **three levels of infection control:** engineering controls, administrative controls and personal protective measures. These levels of control can easily be applied to veterinary practices as well.

1. **Engineering controls** are built into the design of a facility (e.g. room design, sink placement, heating ventilation and air conditioning (HVAC) systems). It is important for infection prevention and control professionals to be involved in the design and planning of new facilities. They can also help to plan and design improvements which may be incorporated into an existing facility. Engineering controls include logical design of clinics to facilitate use of routine infection control measures such as hand washing, proper cleaning, and separation of animals of different species and different infectious disease risks. All new building or renovation plans need to be evaluated from an infection control perspective.

2. **Administrative controls** include protocols for hand hygiene, immunization of animals and staff, protocols for managing animals and staff during an infectious disease outbreak, and protocols for caring for animals with zoonotic infections.

3. **Personal protective equipment (PPE)**, although very important, is the least desirable way to control infectious hazards because it does not eliminate them – it merely contains the hazard. Nonetheless, the inherent risk of exposure to microbial pathogens in veterinary clinics means that proper use of PPE is a critical component of a complete infection control program. Effective use of PPE is dependent on appropriate education and compliance of all staff. Personal protective equipment should be considered a last line of defense for hazards that cannot be overcome with other preventative measures.

The Infection Prevention and Control Program

Every veterinary clinic, regardless of type or size, should have a formal infection prevention and control program that is coordinated by one specific person. This **infection control practitioner (ICP)** should develop protocols, ensure that protocols are being followed, act as a resource for infection control questions, ensure proper training of new staff, direct and interpret surveillance and communicate with staff regarding infection control issues.

The ICP is not necessarily a cumbersome or time-consuming job, as many may think! The day-to-day responsibilities are typically minimal. It is also a not a position that needs to be filled by an expert in infection control or someone with specific training, although that is certainly desirable. In human hospitals, ICPs are typically nurses with specialized infection control training, who perform the day-to-day infection control duties and work under an infection control head, who is typically a physician with training in one or more of infection control, infectious diseases, microbiology and/or public health. These individuals are rarely available in veterinary medicine, but that does not mean that an effective program cannot be established. Either a veterinary technician or veterinarian would be an appropriate ICP in a veterinary clinic. Formal training would be ideal, but if not readily available the key requirement for the position is an interest in infection control. Ideally, over time, the ICP will advance his or her skills through formal and informal continuing education.

In veterinary clinics, the ICP should be the central infection control resource. Among other duties, he or she should:

- Help facilitate development of a written infection control manual.
- Direct and document training of new staff (particularly lay staff).
- Perform formal or informal quality control evaluation of infection control practice compliance (e.g. observing cleaning and disinfection practices, hand hygiene).
- Be the person designated to receive information about and record incidents of suspected hospital-associated infections.

A **written infection control manual** is a critical part of the infection control program. Written documentation can clearly explain infection control practices, ensure that new staff members are properly informed and raise awareness about infection control. Furthermore, written documentation may be important legally in the event of hospital-associated, or more concerningly, zoonotic infections. A written manual demonstrates a level of awareness and effort towards infection control and could be a critical measure to reduce liability risks by demonstrating use of some degree of due diligence.

Support of hospital administration is also crucial to an effective infection control program. If practice owners and managers are unwilling to provide the ICP with adequate time, resources and support, the infection control program will fail. Hospital administration needs to ensure that all veterinary personnel understand and accept the importance of an infection control program, and intervene when required if issues (e.g. poor compliance) arise.

Every veterinary clinic, regardless of type or size, should have a formal infection prevention and control program that is coordinated by one specific person.

Surveillance

Effective infection control is impossible without surveillance, and some form of surveillance should be routinely practiced by all veterinary facilities. Surveillance is the ongoing, systematic collection, analysis, and interpretation of health events with the primary goal being the implementation of response procedures to control adverse outcomes (Burgess & Morley 2015). It is a key component of any infection control program. **Many clinical aspects of surveillance are easy, inexpensive and can be readily incorporated into day-to-day veterinary practice.**

Surveillance efforts are important for establishing and detecting changes in baseline rates of infections within a practice or patient population. If surveillance indicates an increased rate of infection (e.g. increased surgical site infections (SSIs)) this should trigger an investigation into the cause (e.g. patient preparation technique, disinfectant solutions, sterilization equipment, intra-operative techniques, post-operative management), and if appropriate one or more interventions to address the cause(s)(also see SSI surveillance section in Chapter: Surgery for more information). Surveillance can also help assess the effectiveness of these and other interventions or changes in procedures (e.g. use of a new disinfectant).

Every clinic should strive to implement surveillance practices as a part of their infection prevention and control program. The size and scope of such a program will be dependent on clinic size and case load, and should be tailored to the individual clinic's needs. Some guidelines and considerations to help clinics develop their own surveillance program are provided below.

Considerations for clinic infection surveillance programs (adapted from Burgess & Morley 2015):

- **Objectives:** Determine what you hope to achieve with the surveillance program (e.g. measure rates of surgical site infections (SSIs) and compare to baseline).
- **Measured outcomes:** Determine what you will actually measure. For instance, measuring "SSIs" is not defined enough. The specific components of the diagnosis need to be described. (e.g. an SSI occurs when there is inflammation, including redness, swelling and heat, with purulent discharge and/or a positive bacterial culture within 30 days after surgery).
- **Monitored group(s):** Decide who will be monitored for the development of the outcome (e.g. all surgical patients or only those undergoing a specific procedure).
- **Methods (data collection, time, method):** The type of surveillance that will be used should be defined. This could be active surveillance, such as a specific follow-up survey sent to owners of animals that have recently had surgery, or passive surveillance through appointments for suture removal that are scheduled regardless.
- **Critical limit (when action should be taken):** The goal of a surveillance program is to identify problems, if they exist, and take actions to ameliorate the situation when necessary. The level at which action will be taken should be defined (e.g. when the SSI rate exceeds a predetermined level, further investigation and a deep cleaning of all surgical surfaces will be conducted).
- **Reporting mechanism:** The mechanism of reporting to hospital personnel should be predetermined (e.g. monthly report).
- **Document:** The surveillance plan should be documented in writing for easy reference.

Passive surveillance

In the absence of an ongoing infectious disease outbreak, **passive infectious disease surveillance is likely adequate for most clinics**. Passive surveillance is practical, cost-effective and can be performed in any clinic. It involves analysis of data that are already available (e.g. bacterial culture and susceptibility results, results of other kinds of infectious disease testing) to characterize elements such as endemic disease rates, antimicrobial susceptibility patterns and trends, and changes in disease patterns. An example of passive surveillance would be monitoring clinical signs of surgical site infection (SSI) (as opposed to actively collecting culture samples) following all surgical procedures and specific surgical procedures (e.g. spays, neuters). Monitoring of bacterial culture and susceptibility testing can provide information regarding possible outbreaks of hospital associated infections (HAIs), as well as information to guide empirical antimicrobial therapy. Routine recording of animals with specific syndromes such as vomiting, diarrhea, coughing or sneezing is another simple means of providing information that can help in the prevention and early detection of outbreaks, and can help to identify index cases should a hospital outbreak occur.

Post-discharge surveillance is more difficult, but is very important for conditions such as SSIs, as many such infections do not develop until after the animal is discharged from the hospital. Post-discharge surveillance can consist of direct examination of the patient during a recheck appointment, evaluation of readmission data or simply telephone or email contact with owners as part of routine follow up.

The keys to passive surveillance are to establish a consistent case definition so results can be compared over time, to centralize the available data, and to have a designated ICP who compiles, evaluates and reports on the data on a regular basis. **Simply collecting the data or even entering it in a spreadsheet is of limited value unless someone evaluates it and makes a plan for action depending on the results.** This is particularly important in large clinics or hospitals where multiple veterinarians may have patients with similar infections but do not communicate this to others, and therefore the start of an outbreak can be missed. If an outbreak is identified early, then a plan can be formulated and implemented in order to stop the spread of disease. This plan may include additional active surveillance to identify additional cases.

The keys to passive surveillance are to establish a consistent case definition so results can be compared over time, to centralize the available data, and to have a designated ICP who compiles, evaluates and reports on the data on a regular basis.

Active surveillance

Active surveillance involves gathering data for a specific purpose, in this case for informing an infection control program. As a result, this kind of surveillance is usually more expensive and time consuming but usually provides the highest quality data. This is rarely needed in most veterinary clinics and is typically reserved for large facilities with increased infection control threats and personnel available to direct such testing, or during a specific outbreak investigation. An example of active surveillance is collection of nasal and rectal swabs from all animals being admitted to a hospital, whether or not they have signs of infection, to screen for methicillin-resistant *Staphylococcus aureus* (MRSA). Surveillance can be blanket (include all patients), targeted (only include patients in a particular high-risk group), or pulsed (only include patients during a certain period such as a specific day of the week or week within a month). Targeted and pulsed strategies can help decrease time and costs required for surveillance to fit within available resources.

Syndromic surveillance

Syndromic surveillance relies on visible signs of infection or inflammation that can be observed before confirmation from a laboratory test. As such, it has the advantages of being easy (even for support staff), immediate and essentially free to perform. It is recommended in most human hospitals and health care systems as a method of early detection of disease (PHO 2014). Syndromic surveillance is particularly useful for detecting potential outbreaks of conditions such as respiratory or gastrointestinal infections. This allows staff and veterinarians to implement appropriate infection control measures sooner to reduce the risk of transmission of infectious agents. It can also help staff members with triaging patients when booking appointments or upon arrival to the clinic, in order to detect high-risk animals before transmission within the clinic occurs.

Information to assess when booking appointments for sick patients
(adapted from Guptil 2015, see below)

1. Age of patient
2. Vaccination history
3. Recent history (e.g. within the last month):
    a. travel within or outside of province/state/country (or contact with dogs that have done so)
    b. dog park visits
    c. boarding kennel or daycare facility visits
    d. groomer visits
    e. contact with other sick pets
4. Signs of any of these syndromes:
    a. vomiting
    b. diarrhea (3+ loose stools in 24 hours)
    c. coughing
    d. sneezing
    e. lethargy, including lack of appetite (consistent with flu-like illness)

If answers to some or all of questions 4a-4e are "yes," staff should make a note in the appointment calendar to follow clinic protocols for infectious cases (see Chapter: Hospital Associated Infections and Other Infectious Syndromes). Unvaccinated animals, those less than 2 years of age, and those with exposure to other animals (question 3) are at even greater risk of carrying an infectious disease.

References

Burgess BA, Morley PS. Veterinary hospital surveillance. Vet Clinics North Am Small Anim Pract. 2015;45:235-242.

Guptill L. Patient Management. Vet Clinics North Am Small Anim Pract. 2015;45:277–298.

Public Health of Ontario (PHO). Provincial Infectious Diseases Advisory Committee (PIDAC). Best practices for surveillance of health care-associated infections in patient and resident populations. 3rd ed. Toronto, ON: Queen's Printer for Ontario, 2014. Available at: https://www.publichealthontario.ca/en/eRepository/Surveillance_3-3_ENGLISH_2011-10-28%20FINAL.pdf. Accessed Dec-2018.

Antimicrobial Stewardship

A growing issue in both the veterinary and human medical professions is the continuing emergence of antimicrobial resistance (AMR). The challenges presented by multi-drug resistant (MDR) bacterial infections are numerous and affect animals and humans alike. Treating these infections can be time consuming, costly for clients, and may be impossible in some cases, ultimately leading to euthanasia. It is critical for the safety of patients, veterinary personnel and clients to prevent and reduce AMR in clinics. Although this is a multifaceted problem, one of the drivers of AMR is overuse of antimicrobials. While the focus in the past has primarily been on food-producing animals and agriculture, more attention is now being paid to companion animal practices (Prescott & Boerlin 2016). Many of the commonly used antimicrobials in companion animal medicine are also considered to be of high importance in human medicine, and practitioners are urged to exercise caution regarding their use. However, the solution to AMR is not as simple as just reducing antimicrobial use. A balance must be met between prudent antimicrobial use, effective treatment of sick animals, infection prevention and control measures, welfare concerns, public health and the human-animal bond. As such, it is important for clinics to develop antimicrobial stewardship practices, which can be formalized into an Antimicrobial Stewardship Program (ASP).

Antimicrobial stewardship refers to "the multifaceted approaches required to sustain the efficacy of antimicrobials and minimize the emergence of AMR." (Weese 2013) While a complete list of the components of an ASP are beyond the scope of this document, infection prevention and control plays a critical role in any such program. These practices help reduce MDR pathogens within the clinic environment, thereby reducing their potential to infect patients or veterinary personnel and the subsequent need for antimicrobial treatment. The best practices recommended in this document should be applied as part of an ASP.

Recommendations and guidelines are progressively being developed to help small animal clinics implement ASPs, including the BSAVA PROTECT ME program, ISCAID antimicrobial use guidelines, and the ACVIM consensus statements on therapeutic antimicrobial use in animal (see References below). Clinics should also be familiar with jurisdictional and/or national antimicrobial use regulations (if they exist) when developing an ASP to ensure they are in compliance with any such regulations.

Possible components of a clinic antimicrobial stewardship program, in addition to infection prevention and control policies (for additional information, see References):

- Checklists
- De-escalation of therapy
- Antimicrobial time-outs
- Automatic stop orders
- Timely sample collection
- Cascading susceptibility results
- Formulary restriction
- Targeted patient review
- Prescriber education
- Computerized decision-making support

References

British Small Animal Veterinary Association (BSAVA). PROTECT ME. 2019. Available at: https://www.bsava.com/Resources/Veterinary-resources/PROTECT-ME. Accessed Dec-2018.

Hillier A, et al. Guidelines for the diagnosis and antimicrobial therapy of canine superficial bacterial folliculitis (Antimicrobial Guidelines Working Group of the International Society for Companion Animal Infectious Diseases). Vet Dermatol. 2014;25(3):163-e43. PubMed PMID: 24720433.

Lappin MR, et al. Antimicrobial use guidelines for treatment of respiratory tract disease in dogs and cats: Antimicrobial Guidelines Working Group of the International Society for Companion Animal Infectious Diseases. J Vet Intern Med. 2017;31(2):279-294. PubMed PMID: 28185306.

Morley PS, et al. ACVIM consensus statement: Antimicrobial drug use in veterinary medicine. J Vet Intern Med. 2005;19:617-29. Available at: https://onlinelibrary.wiley.com/doi/pdf/10.1111/j.1939-1676.2005.tb02739.x. Accessed Dec-2018.

Prescott JF, Boerlin P. Antimicrobial use in companion animals and good stewardship practice. Vet Rec. 2016;179:486-488.

Weese JS, et al. Antimicrobial stewardship in animals. In: Antimicrobial Therapy in Veterinary Medicine. 5th ed. Giguere S, Prescott JF, Dowling PM (eds). Ames, Iowa: Wiley Blackwell; 2013.

Weese JS, et al. ACVIM consensus statement on therapeutic antimicrobial use in animals and antimicrobial resistance. J Vet Intern Med 2015;29:487-98. Available at: https://onlinelibrary.wiley.com/doi/abs/10.1111/jvim.12562. Accessed Dec-2018.

Weese JS, et al. International Society for Companion Animal Infectious Diseases (ISCAID) guidelines for the diagnosis and management of bacterial urinary tract infections in dogs and cats. Vet J. 2019;247:8-25. PubMed PMID: 30971357.

Routine Practices

Hand Hygiene

Hand hygiene is the responsibility of all individuals involved in health care. It is one of the easiest and most effective ways to prevent infections in the healthcare setting. Effective hand hygiene kills or removes microorganisms on the skin while maintaining hand health and skin integrity (i.e. prevents chapping and cracking of skin). Sterilization of the hands is not the goal of routine hand hygiene – the objective is to reduce the number of microorganisms on the hands, particularly the number of microorganisms that are part of the transient microflora of the skin, as these include the majority of opportunistic pathogens on the hands. These transient microbes may be picked up by contact with a patient, another person, contaminated equipment, or the environment. There are two main methods of removing/killing microorganisms on hands: washing with soap and running water or using an alcohol-based hand sanitizer.

Hand hygiene is one of the easiest and most effective ways to prevent infections in the healthcare setting.

When to perform hand hygiene

- Before and after contact with a patient
  - Especially before performing invasive procedures
- Before and after contact with items in the patient's immediate environment
- Before any aseptic or invasive procedure
- Before putting on and especially after taking off gloves
- After any contact with or any activity involving the body fluids of a patient
- Before eating food or having any hand-to-mouth contact
- After personal body functions, such as using the toilet or blowing one's nose

Factors that influence the effectiveness of hand hygiene

- **Condition of the skin:** Intact skin is easier to clean than skin that is chapped, cracked, cut, abraded or otherwise inflamed. Appropriate use of moisturizers as needed should be encouraged to maintain good skin condition (see skin care section).
- **Finger nails:** Natural nails more than 3-4 mm long are difficult to clean, can pierce gloves and harbour more microorganisms than short nails. Artificial nails or nail extenders should not be worn by anyone involved directly in patient care, as they have been implicated in the transfer of microorganisms in human medicine. Chipped nail polish may also increase the number of microbes found on fingernails (Boyce 2002).
- **Jewelry:** Jewelry can be very hard to clean, and physically protects bacteria and viruses from the antiseptic action of alcohol-based hand sanitizers and the mechanical cleaning action of soap and running water. Rings, in particular, increase the number of microorganisms present on hands and increase the risk of tears in gloves. Hand and arm jewelry (e.g. rings, bracelets) should be avoided as much as possible even for routine patient care, but at a minimum should be removed before aseptic procedures to facilitate effective hand hygiene.

Hand washing with soap and water

Most transient bacteria present on the hands are removed during the mechanical action of washing, rinsing and drying hands. Hand washing with soap and running water must be performed when hands are visibly soiled. If running water is not available, use moistened towelettes to remove all visible dirt and debris, followed by an alcohol-based hand sanitizer.

Bar soaps are not acceptable in veterinary practice settings because of the potential for indirect transmission of pathogens from one person to another due to contamination of the bar itself. Instead, liquid or foam soap should be used with the following recommendations:

- Use a disposable pump dispenser for soap.
- Do not refill soap containers without first cleaning and disinfecting them, since there is a risk of contamination of the bottle itself which increases over time and with use. Cleaning should include scrubbing with a detergent to break up biofilms that may have formed. Use of non-refillable bottles is recommended (PHO 2014).
- **Use antibacterial soaps in critical care areas** such as ICU, and in other areas where invasive procedures are performed. Non-antibacterial soaps are adequate for areas such as washrooms and kitchen areas.

Hand washing technique (see Boyce 2002 and Longtin 2011 for additional information and instructional video):

1. Remove all hand and arm jewelry.
2. Wet hands with warm (not hot) water. Hot water is hard on the skin, and will lead to dryness and additional skin damage.
3. Apply liquid or foam soap to one palm.
4. Vigorously lather all surfaces of both hands for a **minimum of 15 seconds.** This is the minimum amount of time required for mechanical removal of transient bacteria. Pay particular attention to finger tips, between fingers, backs of the hands and base of the thumbs. These are the most commonly missed areas. A simple way many people time their hand-washing is by singing "Happy Birthday" twice.
5. Using a rubbing motion, thoroughly rinse soap from hands under warm running water. Residual soap on the skin can lead to dryness and cracking of skin.
6. Dry hands thoroughly by blotting gently with a paper towel. Rubbing vigorously with paper towels can damage the skin.
7. Turn off taps using the paper towel to protect the hands and avoid recontamination of the hands.

NOTE: If air hand dryers are used, hands-free taps are necessary, as turning taps off without using paper towel as described will result in recontamination of hands after washing.

Alcohol-based hand sanitizers

Alcohol-based hand sanitizers/rubs are, with some exceptions, the preferred method for decontaminating hands that are not visibly soiled. They have superior ability to kill microorganisms on the skin than even hand washing with antibacterial soap, can quickly be applied, are less likely to cause skin damage, and can be made readily available at almost any point of care. Use of non-alcohol-based waterless hand sanitizers in healthcare settings is not recommended.

Alcohol-based hand sanitizers should contain 70-90% alcohol. Use of products containing emollients helps to reduce skin damage which can otherwise occur with frequent use of hand sanitizers. They may be more useful as alternatives to traditional surgical scrubbing techniques (see Chapter: Surgery).

Alcohol-based hand sanitizers are not effective against certain pathogens, including bacterial spores (e.g. clostridial spores) and some protozoa (e.g. Cryptosporidium spp.). Alcohol is also not as effective against non-enveloped viruses (e.g. canine parvovirus, feline panleukopenia virus, feline calicivirus) as it is against most other microbes. Nonetheless, alcohol-based hand sanitizers may be useful even if alcohol-resistant pathogens are present. The improved hand hygiene compliance seen with alcohol-based hand sanitizers and their efficacy against other pathogens are important aspects of infection control. However, if hands are potentially contaminated by an alcohol-resistant organism, hand washing with soap and running water should be performed if possible. Although antimicrobial soaps are similarly ineffective against these pathogens directly, the physical process and mechanical action of hand washing can decrease the number of these organisms on the hands.

Hand sanitizer technique:

1. Remove all hand and arm jewelry.
2. Ensure hands are visibly clean (if soiled, follow hand washing steps).
3. Apply between 1 to 2 full pumps or a 2-3 cm diameter pool of the product onto one palm.

4. Spread the product over all surfaces of hands, concentrating on finger tips, between fingers, back of the hands, and base of the thumbs. These are the most commonly missed areas.
5. Rub hands until product is **dry**. This will take a **minimum of 15 to 20 seconds** if sufficient product is used.
    - Hands must be fully dry before touching the patient or patient's environment/equipment for the sanitizer to be effective, and to eliminate the rare risk of flammability in the presence of an oxygen-enriched environment, as may occur in the presence of gas anesthetic machines.

Intact skin is the first line of defense against bacteria.

Skin care

Careful attention to skin care is an essential part of a hand hygiene program. Intact skin is the first line of defense against bacteria. Products used for hygiene should be "hand-friendly" — for example, alcohol-based hand sanitizers containing emollients can help reduce the drying effect of the alcohol. If skin integrity is an issue, the individual should consult his or her physician. Skin lotions can help maintain the health and integrity of the skin, but it is important to use a skin lotion that does not interfere with glove integrity or antiseptic efficacy. Petroleum or oil-based lotion formulations can weaken latex gloves and increase permeability, therefore these types of products should only be used at the end of the work day (Kohn 2003). If lotions are used during the work day, select a water-based product. Consideration should be given to product selection to ensure there are no negative interactions between gloves or antiseptic agents and lotions.

Compliance

Compliance with hand hygiene protocols is the most challenging component of ensuring efficacy. Numerous studies have investigated hand hygiene compliance in human hospitals (Boyce & Pittet 2002; Larson 2007) and, in general, compliance is poor (<50%) among healthcare workers. In one study, video observation of hand hygiene among veterinary staff was shown to be only 14% in terms of frequency, and duration was considered inadequate in the vast majority of attempts (Anderson 2014).

Major barriers to compliance in many situations include skin irritation (irritant contact dermatitis), lack of accessible hand hygiene stations, time constraints (i.e. too busy), lack of perceived importance relative to other duties, and forgetting to perform hand hygiene activities (Anderson & Weese 2016).

Improving compliance

- Providing **better access** to hand hygiene stations and supplies minimizes the time required to comply with protocols, and acts as a visual reminder to perform hand hygiene. Because renovating facilities to improve the location of sinks for hand washing is often not feasible, improving access typically involves introducing or changing the location and/or number of alcohol-based hand sanitizer dispensers, e.g. placing one by the door inside each exam room.
    - Be aware of potential local fire code regulations regarding installation of wall dispensers near electrical outlets or in carpeted areas.
- **Monitoring and feedback systems** can also have a significant impact on compliance. Feedback may be from fellow staff, patient owners, researchers or infection control personnel. Making people aware of how often they neglect to perform hand hygiene, and/or giving them positive feedback when their compliance improves (the "stick and carrot" approach), seems to provide additional incentive to further improve compliance. Just as hand hygiene compliance among physicians is often lower than among nurses (Pittet 2001, Pittet 1999), the same may be true of veterinarians and technicians/nurses in some cases (Smith 2013). Empowering technicians to remind veterinarians to perform hand hygiene and observe other infection control protocols may help to improve overall compliance.
- **Convince staff** of the importance and utility of hand hygiene in curbing the spread of infectious agents. Staff generally recognize the importance of hand hygiene and the appropriate times to perform it, yet compliance remains low. It may be beneficial to highlight that hand hygiene is also a critical means of preventing potential indirect transmission of non-zoonotic pathogens in addition to those that are zoonotic, from animal-to-animal, thus protecting veterinary patients.

- Make hand hygiene a **team effort**. In order to be effective, infection control measures such as hand hygiene need to be practiced by every member of the clinic team, from veterinarians and technicians to kennel staff and volunteers. Several studies have suggested that involvement and support of upper-level management and administration are necessary for effective implementation of hand hygiene protocols in human health care facilities (Whitby 2007) and it is likely that the same is true in veterinary clinics.

It is reassuring to the client to see clinic personnel performing hand hygiene, particularly within the exam room, and it increases client awareness of the importance of hand hygiene. Practices may wish to reinforce this further by providing alcohol-based hand sanitizers in the waiting room as well.

References

Anderson ME, Sargeant JM, Weese JS. Video observation of hand hygiene practices during routine companion animal appointments and the effect of a poster intervention on hand hygiene compliance. BMC Vet Res. 2014;10:106. PubMed PMID: 24885304.

Anderson ME, Weese JS. Self-reported hand hygiene perceptions and barriers among companion animal veterinary clinic personnel in Ontario, Canada. Can Vet J. 2016;57(3):282-8. PubMed PMID: 26933265.

Boyce JM, et al. Guideline for hand hygiene in health-care settings. Recommendations of the Healthcare Infection Control Practices Advisory Committee and the HICPAC/SHEA/APIC/IDSA Hand Hygiene Task Force. MMWR Recomm Rep. 2002;51(RR-16):1-45. PubMed PMID: 12418624. Available at: https://www.cdc.gov/mmwr/PDF/rr/rr5116.pdf. Accessed Dec-2018.

Kohn WG, et al. Guidelines for infection control in dental health-care settings--2003. MMWR Recomm Rep. 2003 Dec 19;52(RR-17):1-61. PubMed PMID: 14685139. Available at: https://www.cdc.gov/mmwr/preview/mmwrhtml/rr5217a1.htm. Accessed Dec-2018.

Kretzer EK, Larson EL. Behavioral interventions to improve infection control practices. Am J Infect Control. 1998;26(3):245–53.

Larson EL, Kretzer EK. Compliance with handwashing and barrier precautions. J Hosp Infect. 1995;30 Suppl:88–106.

Larson EL, et al. Dissemination of the CDC's Hand Hygiene Guideline and impact on infection rates. Am J Infect Control. 2007;35(10):666–75.

Longtin Y, et al. Videos in clinical medicine. Hand hygiene. N Engl J Med. 2011;364:e24. Available at: https://www.nejm.org/doi/full/10.1056/NEJMvcm0903599. Accessed Dec-2018.

Pittet D. Improving adherence to hand hygiene practice: a multidisciplinary approach. Emerg Infect Dis. 2001;7(2):234–40.

Pittet D, et al. Effectiveness of a hospital-wide programme to improve compliance with hand hygiene. Lancet. 2000;356(9238):1307–12.

Pittet D, et al. Compliance with handwashing in a teaching hospital. Ann Intern Med. 1999;130(2):126–30.

Public Health Ontario (PHO). Provincial Infectious Diseases Advisory Committee (PIDAC). Best practices for hand hygiene in all health care settings. 4th ed. Toronto, ON: Queen's Printer for Ontario; 2014. Available at: https://www.publichealthontario.ca/en/eRepository/2010-12%20BP%20Hand%20Hygiene.pdf. Accessed Dec-2018.

Smith JR, Packman ZR, Hofmeister EH. Multimodal evaluation of the effectiveness of a hand hygiene educational campaign at a small animal veterinary teaching hospital. J Am Vet Med Assoc. 2013;243(7):1042–8.

Whitby M, et al. Behavioural considerations for hand hygiene practices: the basic building blocks. J Hosp Infect. 2007;65(1):1–8.

It is reassuring to the client to see clinic personnel performing hand hygiene

Personal Protective Equipment (PPE)

Personal protective equipment (PPE), including dedicated hospital attire, is an important routine infection control tool. Use of PPE reduces the risk of contamination of personal clothing, exposure of skin and mucous membranes of clinic personnel to pathogens, and transmission of pathogens between patients by staff. Use of PPE does not eliminate the need for appropriate environmental controls, such as hazard removal and separation of patient areas from staff rooms (see Chapter: Basic Principles of Infection Prevention and Control).

Some form of PPE must be worn in all clinical situations, including any contact with animals or their environment. Tables 1 and 2 summarize recommended PPE for routine veterinary procedures, and infectious disease control precautions by disease condition and agent, respectively. These recommendations must always be tempered by professional judgment, while bearing in mind the basic principles of infectious disease control, as every situation is unique in terms of the specific clinic, animal, personnel, procedures and suspected infectious disease.

Use of PPE does not eliminate the need for appropriate environmental controls, such as hazard removal and separation of patient areas from staff rooms.

Personal protective outerwear

Personal protective outerwear is used to reduce the risk of pathogen transmission by clothing to patients, owners, other personnel and the public. It also allows for easy removal of the outer layer of clothing should it become contaminated. Street clothes should be covered by protective outerwear of some kind, such as a lab coat, whenever there may be contact with an animal or when working in a clinical environment (including cleaning). Personal protective outerwear, including lab coats, scrubs and other dedicated hospital attire, should not be worn outside of the work environment, in order to prevent transmission of pathogens between the clinic and household / public places. These items should not be taken home by personnel to be laundered, rather they should be washed on-site, along with other clinic laundry.

Lab coats

Lab coats are meant to protect clothing from contamination, but generally they are not fluid resistant, so they should not be used in situations where splashing or soaking with potentially infectious liquids is anticipated. Lab coats worn during patient handling should be removed prior to performing clean tasks, such as eating and reprocessing equipment. They should be changed promptly whenever they become visibly soiled or contaminated with body fluids, and at the end of each day. When handling patients with potentially infectious diseases, lab coats should be laundered after each use, because it is almost impossible to remove, store/hang and reuse a contaminated lab coat without contaminating hands, clothing or the environment.

Scrubs

Short-sleeved scrub tops and scrub pants are often worn in veterinary clinics as a form of basic personal protective outerwear. They have the advantage of being durable and easy to clean, and their use prevents contamination and soiling of street clothes worn by personnel outside the clinic. Like lab coats, they are not fluid resistant, and they should be changed promptly whenever they become visibly soiled or contaminated with body fluids, and at the end of each day.

Designated scrubs should always be worn during surgery — these scrubs should not be worn during other procedures or when handling patients outside of surgery. Scrubs worn for surgery should be covered with a lab coat outside of the surgical area.

Street clothes should be covered by protective outerwear of some kind, such as a lab coat, whenever there may be contact with an animal or when working in a clinical environment. Personal protective outerwear, including lab coats, scrubs and other dedicated hospital attire, should not be worn outside of the work environment.

Non-sterile gowns

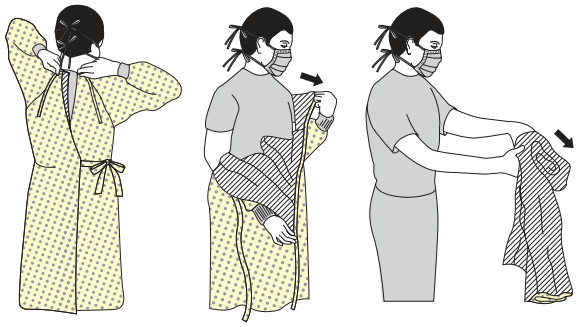
Gowns provide more coverage for barrier protection than lab coats, and are typically used for handling animals with suspected or confirmed infectious diseases, including those housed in isolation. Gowns should cover the torso and arms, and fit snugly at the wrist. Permeable gowns can be used for general care of patients in isolation. Use impermeable (i.e. waterproof) gowns to provide greater protection when splashes or large quantities of body fluids are present or anticipated. Do not reuse disposable gowns. Launder reusable fabric gowns after each use, because hanging/storing and reusing contaminated gowns inevitably leads to contamination of hands, clothing or the environment. Wear gloves whenever gowns are worn. Gowns (and gloves) should be removed and placed in the trash or laundry bin before leaving the animal's environment, and hand hygiene should be performed immediately after.

It is important to train personnel on how to remove (doff) gowns properly, in such a way as to avoid contaminating themselves and the environment. The outer (contaminated) surface of a gown should only be touched with gloves.

Procedure for doffing of personal protective equipment (modified from PHO 2012, CDC 2016):

1. **Disposable gown:** With gloves still on, grasp the front of the gown and pull it away from the body, breaking the ties, and fold or roll the gown inside-out into a bundle as it is removed. As the sleeves are removed, also peel off gloves leaving them inside the bundle. Only touch the inside of the gloves and gown with bare hands. Place the bundle directly in a waste container.
2. **Reusable gown:** Remove gloves first. Unfasten or break ties behind the neck. Peel the gown down from the shoulders and arms, only touching the neck ties and inside surfaces of the gown with bare hands. Fold or roll the gown inside-out into a bundle as it is removed. Place the bundle directly in a waste container or laundry bin.
3. Perform hand hygiene.
4. Remove eye protection (if applicable).
5. Remove mask (if applicable).
6. Perform hand hygiene.
7. If body fluids soaked through the gown, promptly remove the contaminated underlying clothing and wash the skin.

FIGURE 1. How to remove a gown



https://www.cdc.gov/hai/pdfs/ppe/PPE-Sequence.pdf

Launder reusable fabric gowns after each use, because hanging/storing and reusing contaminated gowns inevitably leads to contamination of hands, clothing or the environment.

Additional personal protective equipment

Gloves

Gloves reduce the risk of pathogen transmission by providing a protective barrier to bare hands. They should be worn when contact with blood, body fluids, secretions, excretions and mucous membranes is anticipated. Gloves should also be worn when cleaning cages and environmental surfaces, and when handling dirty laundry (especially if gross contamination of items is present) Gloves are NOT a substitute for proper hand hygiene. It is a common misconception that using disposable gloves negates the need for hand hygiene. Gloves do not provide complete protection against hand contamination, therefore hand hygiene immediately after removing gloves is essential (Korniewicz 2004).

Proper glove use includes the following:

- Remove gloves promptly after use, avoiding contact between skin and the outer glove surface.
- Do not touch surfaces with gloved hands that will be touched by people with non-gloved hands.
- Take care to avoid touching and contaminating personal items such as telephones, pens and pagers.
- Do not touch one's face or glasses with gloved hands.
- Wash hands or use an alcohol-based hand sanitizer immediately after glove removal.
- Do not wash or reuse disposable gloves.

Gloves are NOT a substitute for proper hand hygiene

Change gloves and perform hand hygiene when:

- Moving from contaminated areas to clean areas on the same animal.
- Moving from dirty to clean procedures on the same animal.
- After contact with large amounts of blood and/or body fluids.
- Between individual animals.

Gloves come in a variety of materials. The choice of glove material depends on their intended use. Latex gloves are commonly used, but if latex allergies are a concern, acceptable alternatives include nitrile or vinyl. Latex gloves will decompose and lose their integrity when exposed to many chemicals. If exposure to chemicals such as disinfectants is expected (e.g. when cleaning and disinfecting cages), disposable nitrile gloves or heavier, reusable rubber gloves (e.g. common dishwashing gloves) can be used. Reusable gloves must be disinfected at the end of each task. It is also important to provide a suitable variety of glove sizes to accommodate all staff who are expected to use them, so that they fit properly (not too big or too small).

Face protection

Face protection prevents exposure of the mucous membranes of the eyes, nose and mouth to infectious materials. Face protection typically includes a nose-and-mouth mask (e.g. surgical mask) and goggles, or a full face shield. Face protection should be used whenever exposure to splashes or sprays is likely to occur, including dental procedures, nebulization, and wound lavage. Masks with a flexible nose piece that can be adjusted to the individual user should be used so that the mask fits snugly around the nose and mouth.

Respiratory protection

Respiratory protection is designed to protect the respiratory tract from infectious pathogens transmitted through the air. **The need for this type of protection is limited in veterinary medicine** because there are few relevant airborne or aerosol zoonotic pathogens in companion animals (e.g. influenza virus, plague (Yersinia pestis)), in most regions. The N95-rated disposable particulate respirator is a type of mask that is inexpensive, readily available, easy to use, and provides adequate respiratory protection in most situations. However, individuals need to be fit-tested to ensure proper placement and fitting of an N95 respirator. Special N95 respirators are required for individuals with beards. Surgical masks are not a substitute for N95 respirators.

Footwear

Closed toed, cleanable footwear must be worn at all times in the clinic to reduce the risk of injury from dropped equipment (e.g. scalpels, needles), scratches from being stepped on by dogs, and to protect the feet from contact with potentially infectious substances (e.g. feces, discharges and other body fluids). Footwear should have adequate tread to minimize the potential for slips, trips, and falls.

Designated footwear or disposable shoe covers are required in areas where infectious materials are expected to be present on the floor, in order to prevent their spread to other areas. This is particularly important in veterinary clinics because patients, and sometimes the personnel working with them, have very close contact with the floor. Designated footwear or disposable shoe covers may be required when handling patients with infectious diseases that are kept on the floor (e.g. in a large dog run) or that may contaminate the floor around their kennel (e.g. an animal with severe diarrhea). Such footwear must be removed as the person leaves the contaminated area, and should be immediately disposed of in the garbage (if disposable), or left at the entrance of the contaminated area on the "dirty" side.

TABLE 1. Recommended personal protective equipment for routine veterinary procedures, in addition to standard personal protective outwear (e.g. lab coat or scrubs) worn at all times in clinical areas.

Procedure	Disposal Gloves	Sterile Gloves	Gown / Dedicated Lab Coat	Face Protection[a]	Other/Comment
Bandage change (routine)	+				
Bandage change (infectious)	+		+	(+)	
Crushing pills					Mask only[b]
Dental procedures	+		+	+	
Digital rectal palpation	+				
Draining sterile seroma/hematoma		+			
Expressing anal glands	+				
Fine needle aspirate	+				
Handling soiled laundry	+		+		
Handling stool samples	+				
Handling urine samples	+[c]				
Injections: intramuscular and subcutaneous					
Intranasal or oral *Bordetella* vaccination	+				
Intravenous catheter placement (long term)		+			Central lines and arterial catheters
Intravenous catheter placement (short term)	+				
Lancing abscess	+		+	(+)	
Obstetrical procedures: cats	+		+	+	Q fever risk
Obstetrical procedures: dogs	+				
Oral antimicrobial administration	+[b]				
Oral examination (detailed)	+				
Urinary catheter placement		+			
Venipuncture					
Wound cleaning/debridement (dirty)	+				
Wound cleaning/debridement (clean)		+			
Wound lavage/flushing	+		+	(+)	
Wound suturing		+			

+ PPE recommended; (+) PPE recommended if splash risk is present. [a] Face shield or protective glasses and face mask; [b] Indicated in individuals with sensitivity to the drug; clf leptospirosis is suspected, glove use is strongly recommended, but consider in cases of other urinary tract infections as well

TABLE 2. Infectious disease control precautions by disease condition and agent

Disease Condition	Agent Name	Disease Name	Zoonotic Risk	Bite/Scratch Concern	Environmental Contamination	Arthropod Vector	PPE Protocol			
							Gloves	Gown[a]	Mask[b]	Other
Respiratory Tract Infection	*Bordetella bronchiseptica*	Bordetellosis	(+)		+		+	+		
	Canine influenza virus	Influenza	(+)		+		+	+		
	Feline calicivirus	Calicivirus			+		+	+		
	Feline herpesvirus 1	FVR			+		+	+		
	Francisella tularensis	Tularemia	+	+	+	+	+	+	+	C
	Pasteurella multocida	Pasteurellosis	+	+						P
	Mycobacterium bovis, Mycobacterium tuberculosis	Tuberculous mycobacteria	+		+		+	+	+	
	Canine parainfluenza virus	Parainfluenza			+		+	+		
Diarrhea	*Campylobacter jejuni*	Campylobacteriosis	+		+		+	+		F, S
	Clostridium difficile	*C. difficile* infection	+		+		+	+		F, S
	Cryptosporidium spp.	Cryptosporidiosis	+		+		+	+		F, S
	Giardia spp.	Giardiosis	+		+		+	+		F, S
	Salmonella spp.	Salmonellosis	+		+		+	+		F, S
	Toxoplasma gondii	Toxoplasmosis	+		+		+	+		F, S
	Canine parvovirus	Parvo			+		+	+		F, S
	Feline panleukopenia virus	Panleukopenia			+		+	+		F, S
Neurological Signs	*Listeria monocytogenes*	Listeriosis	+		+		+	+	+	C
	Canine distemper virus	Distemper			+		+	+		
	Rabies virus	Rabies	+	+			+	+	+	C
Skin Condition External Parasites	MRSA	MRSA pyoderma	+	+	+		+	+		F, C
	MRSP	MRSP pyoderma	(+)		?		+	+		F, C
	Fleas	Fleas	+		+		+	+		
	Lice	Pediculosis			+		+	+		
	Mites	Mange	+		+		+	+		
	Ticks	Ticks	+		+		+			L
	Microsporum spp. Trichophyton spp.	Dermatophytosis, Ringworm	+		+		+	+		
	Sporothrix schenkii	Sporotrichosis	+	+			+			F, S, L
Wounds and Abscesses	MRSA	MRSA	+	+	+		+	+		F, C
	MRSP	MRSP	(+)		?		+	+		F, C
	Pasteurella multocida	Pasteurellosis	+	+						P
	VRE	VRE	+		+		+	+		C, S
	Other MDR bacteria	Other MDR bacteria	+				+	+		C
Fever of Unknown Origin / Non-Specific Clinical Signs	*Bartonella spp.*	Cat Scratch Disease	+	+		+				B
	Borrelia burgdorferi	Lyme Disease				+				B
	Brucella canis	Brucellosis	+				+	+	+	
	Chlamydophila psittaci	Psittacosis	+		+		+	+	+	C

TABLE 2. Continued. Infectious disease control precautions by disease condition and agent

Disease Condition	Agent Name	Disease Name	Zoonotic Risk	Bite/Scratch Concern	Environmental Contamination	Arthropod Vector	PPE Protocol			
							Gloves	Gown[a]	Mask[b]	Other
Fever of Unknown Origin / Non-Specific Clinical Signs ... continued	*Coxiella burnetti*	Q fever	+		+		+	+	+	C
	Francisella tularensis	Tularemia	+			+	+	+	+	C
	Leishmania spp.	Leishmaniasis	+			+				B
	Leptospira spp.	Leptospirosis	+		+		+	+		C, S
	Rickettsia rickettsii	RMSF				+				B
	Toxoplasma gondii	Toxoplasmosis	+		+					F
	Canine distemper virus	Distemper			+		+	+		F
	Canine adenovirus 2	Adenovirus			+		+	+		
	Feline leukemia virus	Feline leukemia			+		+	+		
	FIV	FIV			+[c]					
	Rabies virus	Rabies	+	+			+	+	+	C
	Yersinia pestis	Plague	+	+		+	+	+	+	B, C
Intestinal Worms	*Ancylostoma spp.*	Hookworm	+		+					F
	Dipylidium caninum	Tapeworm	+			+[d]				P
	Echinococcus spp.	Hydatid disease	+		+		+	+		F, S
	Taenia spp.	Tapeworm			+					F
	Toxocara spp.	Roundworm	+		+					F

+ Risk exists/PPE required; (+) moderate risk; ? Unknown risk

FIV – feline immunodeficiency virus; **FVR** – feline viral *rhinotracheitis*; **MDR** – multidrug-resistant; **MRSA** – methicillin-resistant *Staphylococcus aureus*; **MRSP** – methicillin-resistant *Staphylococcus pseudintermedius*; **PPE** – personal protective equipment; **RMSF** – rocky mountain spotted fever; **VRE** – vancomycin-resistant *Enterococcus spp.*

[a] Disposable gown or dedicated lab coat; [b] Mask covering the nose and mouth (e.g. surgical mask) and goggles, or full face shield; [c] Environmental contamination by blood; [d] Transmission by ingestion of fleas

B = Prevent direct contact with blood; **C** = Cover broken skin; **F** = Prevent direct contact with feces and transfer of fecal contamination; **L** = Lab coat (non-dedicated) recommended; **P** = Standard PPE only, according to procedure; **S** = Shoe covers recommended if there is possible fecal contamination (or urine contamination for leptospirosis) of the floor in the area where the animal is being kept

Reportable diseases in animals in Canada included in this table:

Brucellosis
Rabies

Notifiable diseases in humans in Canada included in this table:

Brucellosis
Campylobacteriosis
Clostridium difficile infection
Cryptosporidiosis
Giardiosis
Listeriosis
Lyme Disease
Plague
Psittacosis
Rabies
Salmonellosis
Tularemia

See Chapter: Reportable Diseases and Appendix: Management of Rabies Suspects for more information.

References

Centers for Disease Control and Prevention (CDC). Protecting healthcare personnel: Sequence for donning and removing personal protective equipment. 2016. Available at: https://www.cdc.gov/hai/pdfs/ppe/PPE-Sequence.pdf. Accessed Dec-2018.

Public Health Ontario (PHO). Provincial Infectious Diseases Advisory Committee (PIDAC). Routine practices and additional precautions, in all health care settings (Appendix L). 3rd ed. Toronto, ON: Queen's Printer for Ontario; 2012. Available at: https://www.publichealthontario.ca/-/media/documents/rpap-recommeded-ppe-steps.pdf?la=en. Accessed Dec-2018.

Korniewicz DM, et al. Failure rates in nonlatex surgical gloves. Am J Infect Control 2004;32(5):268-273. PubMed PMID: 15292890.

Patient Care and Handling

Isolation facilities

Every veterinary clinic should have a dedicated isolation area for caring for and housing animals with potentially contagious infectious diseases. The size and structure of the isolation facility will vary based on factors such as clinic size, animal species treated and diseases endemic to the area. A proper isolation area should allow for complete physical separation of potentially infectious cases, and have areas for performing routine procedures such as bandage changes, thereby reducing the risk of direct or indirec t transmission to other hospitalized animals or clinic personnel. Ideally, isolation facilities should be in an area that limits traffic into and near the entrance of the room, but with a means of easy monitoring for patient safety (e.g. webcam or window).

If an isolation area was not included in the original physical design of the clinic, a potential alternative in some cases may be to convert an examination room into a dedicated isolation room. The room selected should be in the area of the lowest human and animal traffic possible. The room should be easy to clean and disinfect and emptied of all non-essential equipment. This type of room conversion can be difficult to do effectively depending on the design and layout of the clinic and the room itself. The feasibility of using such a room for isolation of infectious animals must be assessed on a facility-by-facility basis. Depending on the situation, it may be more feasible and beneficial to refer the client to a facility with a more appropriate isolation facility in order to protect the health of the animal, other patients and personnel.

Ventilation should be designed such that movement of air from the isolation room to other areas of the clinic is prevented (i.e. the room should be vented to the outdoors). If this is not readily possible, a HEPA air filtration system should be used for the air leaving the isolation room. HEPA filters need to be replaced on a regular basis (according to manufacturer's instructions), whether for an entire room or for containment/oxygen cages. Although HEPA filter oxygen cages can provide primary containment of an infectious animal, the risk of cross-contamination by staff remains, so the animal should still be housed in the isolation area regardless.

Every veterinary clinic should have a dedicated isolation area for caring for and housing animals with potentially contagious infectious diseases. All items entering an occupied isolation area should be considered contaminated.

Only the equipment and materials needed for the care and treatment of the individual animal should be kept in the isolation room. This may include items such as a designated stethoscope, thermometer, grooming supplies, leash, and muzzle. Supplies of items that will be used for subsequent isolation patients (e.g. packages of bandage material, boxes of needles and syringes) should not be kept in the isolation area. All items entering an occupied isolation area should be considered contaminated and disposed of or thoroughly disinfected after discharge of the patient (e.g. fluid pumps). Items should not be removed from the room except for disposal or laundering (see Chapter: Laundry and Waste Management). Use of disposable articles can minimize the need to take soiled items out of the isolation room.

When the isolation room is in use by an animal with a potentially contagious infectious disease:

- **Use prominent signage** to indicate that the animal may be infectious and outline any additional precautions that need to be taken in addition to routine isolation protocols.
- **Limit access to the isolation room** to the minimum number of essential personnel necessary to provide appropriate patient care.

Personal protective equipment and waste in isolation

All personnel entering an isolation area housing a potentially infectious animal, regardless of whether they plan on having direct contact with the animal, must wear appropriate personal protective clothing. At a minimum, this consists of a clean lab coat or similar item of outerwear that is only worn in the isolation area and disposable examination gloves. Depending on the diagnosis and the mode of transmission of the disease, shoe covers, masks and eye protection may be required when handling an animal in isolation (see Chapter: Personal Protective Equipment). All non-essential items should be left outside the isolation area, including personal cellphones, extra pens, extra clothing, etc.

After handling a patient in an isolation area (CDC 2016, PHO 2012, also see Chapter: Personal Protective Equipment):

- Discard gloves after a single use and perform hand hygiene after gloves are removed.
- Discard gowns (if disposable) after a single use. Launder reusable gowns and lab coats used in isolation after a single use. Immediately place these items in the isolation room garbage or laundry bag once removed. Storing/hanging and reusing a contaminated gown or lab coat inevitably leads to contamination of hands, clothing and the environment.
- Eye/nose/mouth protection may be re-used with the same animal if they are not visibly soiled and can be consistently removed without contamination of the inside of the eye wear/mask or the immediate environment. Nose and mouth masks may be reused only by the same person. Replace eyewear or masks with a clean article if they become contaminated with body fluids such as urine or feces. **Designated personal protective equipment must remain in the isolation room.**

Contaminated items and waste alike should be bagged prior to removal from the isolation area. Laundry and waste bags should be placed in a second "clean" bag to cover the original contaminated bag from isolation. Other items should also be placed in a "clean" bag and immediately taken to the appropriate area for additional cleaning and disinfection. Waste from an isolation room should be treated as potentially infectious and placed immediately in the appropriate disposal bin (see Chapter: Laundry and Waste Management).

Patients in isolation

The decision of whether to house a patient in isolation often needs to be made on a case-by-case basis. While certain diagnoses or syndromes typically warrant isolation (see management of specific infectious syndromes in Chapter: Hospital Associated Infections and Other Infectious Syndromes), the likelihood of shedding of an infectious pathogen, the risk of transmission to other patients or personnel in the clinic, and the ability to manage the patient effectively in isolation all must be considered.

Dogs that are housed in isolation should ideally not be walked nor allowed to urinate or defecate in public areas or areas used by other animals. If a dedicated area for walking is not available and the dog needs to be taken out of the primary isolation area to urinate and defecate, a separate indoor or outdoor run should be designated for each dog in isolation (i.e. if there is more than one animal in isolation, they cannot all use the same run). The run selected should be as far as possible from runs being used by other animals. The dog should be moved **directly** to the run by personnel wearing appropriate personal protective clothing. Moving the animal through other areas of the clinic should be avoided as much as possible. **Carrying the dog or transporting it on a gurney** is ideal in order to minimize the risk of contamination of the floor and clinic environment. The designated run should be prominently labeled and disinfected at least daily and between patients. Prompt removal of feces and other solid waste (e.g. vomitus) is also crucial. (See outdoor elimination areas section in Chapter: Clinic Design).

If a patient being housed in isolation absolutely must be taken elsewhere in the clinic for essential procedures (e.g. radiographs, surgery), if at all possible this should be done the end of the day, or during a time where there is the least animal and personnel movement in the clinic and adequate time to clean and disinfected potentially contaminated areas prior to next use.

- All personnel involved with the procedure must wear appropriate personal protective equipment
- Keep other animals out of the procedure area while in use.
- Thoroughly clean and disinfect the procedure area as soon as the procedure is completed.

Wound care and bandages

Wound infections can be caused by many bacterial pathogens, some of which can be transmitted between animals or between animals and people. One example is methicillin-resistant *Staphylococcus aureus* (MRSA), which can infect both people and animals, but there are a variety of other pathogens of concern. These include both multidrug-resistant and non-resistant strains of bacteria (e.g. *S. aureus, S. pseudintermedius, enterococci)*. Wounds provide a prime site for invasion of opportunistic bacteria such as these. Even wounds that are not known to be infected should be protected from contamination by veterinary personnel and from the environment to reduce the risk of secondary infection, through the use of bandages and/or other routine protocols as appropriate.

To reduce the risk of infection or contamination of the environment from a wound during bandage changes:

- Wear sterile gloves for debridement, treatment and bandaging of deep wounds and those involving vital structures. Clean, non-sterile examination gloves are adequate for these procedures if the wound is superficial. Wash hands thoroughly before and after glove removal.
- **Bandages must be kept dry** to prevent bacterial strike-through. This means keeping the outside of the bandage as dry as possible, and also including sufficient absorbent material in the bandage itself to prevent discharge from the wound from soaking through the bandage. Change the bandage if the outside of it appears wet for any reason.
- **Consider used bandage materials infectious waste.** Place such materials directly in the garbage and not on the floor, examination table or any other surface. The risk of contamination and spread of any pathogen is likely higher for wounds with a large amount of discharge.
- Proximity to the potentially infectious wound during care increases risk of contamination of unused bandage materials. Keep extra materials away from the area when bandage changes are performed, and discard unused materials that may have been contaminated during the procedure (e.g. unfinished rolls).
- Perform wound treatments and bandage changes **in an area that is easily disinfected** (e.g. on an examination table). Perform wound irrigation and lavage in such a way that the fluid used is contained (e.g. in a sink or tub, or with disposable absorbent material). Do NOT change bandages in the kennel/ward area or in a high-traffic area where there is increased risk of cross-contamination of other patients.
- **Wash hands thoroughly** after changing a bandage. Disinfect equipment used for bandage changes (e.g. bandage scissors) between uses.

Additional personal protective equipment (e.g. gown, face protection) should be worn when managing large wounds or those producing a large amount of discharge (splash risk). Particularly if it is difficult to keep the wound completely covered (based on the nature of the wound or the demeanor of the animal), the patient should ideally be housed in isolation. Animals with known MRSA/MRSP or other multidrug-resistant bacterial wound infections are likely to be colonized with these pathogens at other body sites as well (e.g. nose, rectum, intestinal tract), and should therefore be handled with contact precautions and housed in isolation (see Chapter: Hospital Associated Infections and Other Infectious Syndromes).

Feeding of raw meat diets and treats

Raw animal/meat-based diets and treats for cats and dogs often contain a variety of enteropathogens, including *Salmonella spp, Campylobacter spp, Clostridium difficile, Clostridium perfringens*, extended spectrum beta-lactamase (ESBL)-producing Enterobacteriaceae, and enterohemorrhagic strains of *Escherichia coli* such as O157:H7. It has also been shown that animals fed raw meat diets may shed higher levels of *Salmonella* and ESBL Enterobacteriaceae in their feces (Lefebvre 2008).
Raw animal-based products (diets and treats), and feces from animals fed these products, may pose a risk to hospitalized animals and clinic personnel, and may contaminate the hospital environment. Therefore, **a policy against the feeding of raw animal-based products to hospitalized animals should be in place.** Clients who do not wish to have their animal fed a commercial kibble diet can consider cooking the pet's normal diet for the duration of the hospitalization period. Non-diarrheic animals normally fed a raw meat diet do not necessarily need to be housed in isolation, but they should be walked in designated area and their feces considered potentially infectious (and therefore cleaned up immediately).

If it is the opinion of the attending veterinarian that changing an animal's diet from a raw meat diet for the duration of hospitalization would adversely affect the animal's health, then the following guidelines should be followed (as they should be at home):

- Keep raw meat frozen until the day before feeding. Thaw it in the refrigerator on the bottom shelf in a sealed container.
- Promptly discard any uneaten meat in such a way that it will not attract nor be accessible to insects, vermin or other animals. Significant bacterial growth can occur in any meat that is left out at room temperature, even for a short period of time.
- Thoroughly clean and disinfect any items that come in contact with raw meat (e.g. bowls, storage containers) immediately after use (Weese & Rousseau 2008). Bleach is a good choice for this as it has a broad spectrum of activity and is considered food safe, but items must be thoroughly cleaned first and an adequate concentration and contact time must be observed (see Chapter: Cleaning, Disinfection and Sterilization).
- Strongly emphasize hand hygiene after handling raw meat or any items that have been in contact with raw meat.

High risk admissions

Animals from shelters

Humane societies, animal shelters and similar facilities typically contain transient, stressed populations of animals, large numbers of young animals, sick animals and animals of unknown health and vaccination status. As such, they should be considered high risk from an infectious disease standpoint. Animals admitted from these facilities should be managed with additional precautions:

- Examine all animals from such facilities immediately upon arrival. Do not allow them to come in contact with other animals in the waiting/reception area.
- Always house animals from these facilities separately from other patients, if possible. Use of a separate ward, separate area of a ward or leaving empty cages between these animals and other patients are all potential strategies, depending on the degree of separation required for the diseases of primary concern.
- If there is an ongoing outbreak of an infectious disease at the animal shelter, restrict admission of animals from the facility to emergencies only (i.e. no elective procedures). Admit all animals from the facility directly to isolation until the outbreak is resolved.

Patient management considerations for elective procedures for high-risk animals (e.g. spay / neuter of shelter animals):

- All dogs, cats and ferrets must be **currently vaccinated against rabies,** with their first vaccination given at least 2 weeks prior to presentation (i.e. if vaccinated at the minimum on-label age of 12 weeks, animals should be a minimum of 14 weeks of age at presentation).
- All dogs and cats must be up-to-date on other **core vaccinations,** as needed according to geographic region. Animals more than 14 weeks old should have received at least two doses of vaccine at an appropriate interval, with the most recent vaccine administered at least 2 weeks prior to presentation.
- All animals must be **dewormed** with a broad-spectrum anthelmintic approximately 7-10 days prior to admission. This interval can be longer for animals in which ongoing exposure to parasites has been controlled since the last deworming.
- Do not admit animals for elective procedures if they have abnormalities including, but not limited to, fever, oculonasal discharge, coughing/sneezing, diarrhea and potentially infectious skin conditions.
- Depending on the geographic region and time of year, **flea treatment** prior to admission may also be required.

Imported animals

Clinics should be aware of current importation requirements in their jurisdiction, and apply a high degree of scrutiny to imported dogs. These animals can pose a risk to human and animal health through the introduction of non-endemic diseases into the local dog population, and spread of zoonotic pathogens to humans. Regulatory control over importation varies between countries. For example, Canada requires that dogs in most categories have a certificate of health and/or a rabies vaccination certificate, but there are exceptions to even these basic requirements (Anderson 2016, see below). Ideally, imported dogs will have received a formal health examination prior to entry that includes vaccinations, deworming, and any other relevant health tests (e.g. heartworm testing), but it can be difficult to verify that these procedures have been completed effectively due to the variability in veterinary services and products available in some countries. Dogs may be imported directly by the owner or by larger rescues or other organizations. If shelters are importing dogs, the recommendations for management of animals from shelters (see above) are appropriate.

Veterinarians should take extra precautions when handling imported dogs until they can be relatively certain of the dog's health and behaviour status. Management of the disease risk should include vaccination (repeated as necessary), internal and external parasite treatments, and necessary and relevant diagnostic testing (e.g. heartworm). A canine importation checklist for veterinarians is available from the Canadian Veterinary Medical Association (see References below).

References

Anderson MEC, et al. Report of the Canadian National Canine Importation Working Group. 2016. Available at: https://www.canadianveterinarians.net/documents/canadian-canine-importation-working-group-report. Accessed Dec-2018.

Canadian Veterinary Medical Association (CVMA). Veterinarians' Dog Importation Checklist. 2018. Available at https://www.canadianveterinarians.net/documents/veterinarians-dog-importation-checklist. Accessed Dec-2018.

Centers for Disease Control and Prevention (CDC). Protecting healthcare personnel: Sequence for donning and removing personal protective equipment. 2016. Available at: https://www.cdc.gov/hai/pdfs/ppe/PPE-Sequence.pdf. Accessed Dec-2018.

Lefebvre SL, et al. Evaluation of the risks of shedding Salmonellae and other potential pathogens by therapy dogs fed raw diets in Ontario and Alberta. Zoonoses Public Health. 2008;55:470-480.

Public Health Ontario (PHO). Provincial Infectious Diseases Advisory Committee (PIDAC). Routine practices and additional precautions, in all health care settings (Appendix L). 3rd ed. Toronto, ON: Queen's Printer for Ontario; 2012. Available at: https://www.publichealthontario.ca/-/media/documents/rpap-recommeded-ppe-steps.pdf?la=en. Accessed Dec-2018.

Weese JS, Rousseau J. Survival of Salmonella Copenhagen in food bowls following contamination with experimentally inoculated raw meat: effects of time, cleaning, and disinfection. Can Vet J. 2006:47(9):887-889.

Hospital-Associated Infections and Other Infectious Syndromes

Hospital-associated infections (HAIs) are caused by pathogens encountered within the hospital environment. These infections are often further defined as such by their onset after 48 hours or more of hospitalization (while the patient may still be in hospital) or within 30 days of hospital discharge. This definition can result in both over- and under-diagnosis of HAIs, but provides a standard framework to identify and monitor HAIs.

While it may seem straightforward, identification of HAIs can be a challenge because:

- Most veterinary patient hospital stays are relatively short and clinical signs of infection often only become apparent after discharge.
- Animals may develop signs of infection during hospitalization from diseases they were incubating at the time of arrival.

Hospital-associated infections may be caused by bacteria acquired while in the hospital, or from an animal's own microbiota, but in either case a HAI is associated in some way with the hospital environment or factors / procedures (e.g. surgery, catheterization) that occurred during hospitalization.

In general, common types of HAIs include intravascular (IV) catheter-associated infections, urinary tract infections (UTIs), and surgical site infections (SSIs), in addition to infectious respiratory disease and infectious diarrhea (see Management of other infectious syndromes section). While there are specific considerations for each of these HAIs, general infection prevention and control protocols are effective at reducing the risk of all of them:

- **Use hand hygiene and personal protective equipment** to reduce the transmission of pathogens between patients and personnel, and contamination of clothing and equipment (see Chapter: Hand Hygiene and Chapter: Personal Protective Equipment).
- Follow routine practices for **cleaning and disinfection** of equipment and environment including removal of organic debris, selecting a disinfectant based on spectrum of activity and material compatibility, and following the manufacturer's directions for proper use (especially concentration and contact time) (see Chapter: Cleaning, Disinfection, Sterilization).
- Employ **appropriate patient management** strategies for patients known or suspected to be infectious to others. This includes use of isolation areas, separate equipment, and appropriate patient movement (see Chapter: Patient Care and Handling).
- Pay particular attention to infection control precautions for procedures with higher risks of HAIs such as catheterization (both intravenous and urinary) and surgery.
- Separate staff animals and resident animals from patients (see Chapter: Non-Patient Animals).
- Employ **antimicrobial stewardship** strategies (see Chapter: Antimicrobial Stewardship).
- Establish a **surveillance program** to determine benchmarks of infection rates in order to detect any changes that may indicate an HAI outbreak. (see Chapter: Surveillance).
- **Educate and train** veterinary personnel on zoonotic disease risks and infection prevention and control (IPC) strategies. All veterinary personnel should be familiar with their clinic's IPC policies or guidelines (see Chapter: Education).

Contamination of skin preparation materials

Contamination of containers used to hold gauze soaked in chlorhexidine or other biocides for wound care, intravenous catheter placement or surgical preparation have been implicated in HAIs in both human and veterinary medicine (Mathews 1996). Refilling containers of these solutions without proper cleaning and disinfection allows bacteria to become tolerant to the biocides, allowing for serial contamination (see surgical site management section in Chapter: Surgery).

If containers of pre-soaked gauze or other materials are used:

- Empty them daily, and clean, disinfect and dry the containers themselves regularly. Do not "top" up containers or solutions.
- Use stainless steel containers (rather than plastic), as they are more amenable to frequent cleaning and disinfection.

Bloodstream infections

Intravascular catheters

Intravascular catheters (both venous and arterial) provide direct access for opportunistic pathogens to the bloodstream. Inadequate skin preparation, contamination of skin prep solutions, duration of catheterization, catheter material, location of catheter, and infusion with dextrose increase the risk of an infection.

Precautions to help reduce the risk of a bloodstream infection when placing a catheter:

- Choose a site free from skin lesions or other infections, that is easy to monitor, adequately protected from contamination and amenable to catheterization.
- Choose a catheter of appropriate size and material for the given patient and procedure.
- Clip (don't shave) the site prior to catheter placement, avoiding sites that have been clipped for other procedures.
- At a minimum, prepare the catheter site with an antiseptic, allowing the antiseptic to dry before placement.
  - If there is gross contamination of the skin, clean the site with soap and water first.
- Use aseptic technique for placement, wearing clean or sterile gloves for a peripheral intravenous catheter, and sterile gloves for placing central lines or arterial catheters.
- Monitor the catheter for signs of heat, pain, swelling or discharge daily, or more frequently if complications are expected. If necessary, bandage the catheter in a way that makes monitoring possible. Change the bandage if visibly soiled. Bandage movement can be a potential issue for catheter maintenance and in some cases may increase the risk for infections and/or thrombophlebitis
- Minimize direct contact with the catheter site; perform hand hygiene and wear clean, disposable gloves when contact is required.
- Remove the catheter as soon as it is no longer required or is medically indicated. There is no evidence that routine catheter changes are beneficial.

Catheter flushing should be performed using good hygiene practices to prevent inoculation of bacteria. Only sterile fluids should be used for catheter flushing. Individual fluid vials are ideal, because of the potential for contamination of multidose vials or bags after repeated entry. In situations where this is not economically or logistically feasible, care must be taken to reduce the risk of contamination of multidose flush bottles or bags, and these items should be disposed if not used up within a specific time frame (e.g. 24 hours). It is recommended to flush catheters every 4 hours with saline unless fluids are used continuously; use of heparinized saline does not appear to be of any additional benefit (Davis 2013).

Intravenous solutions, including parenteral nutrition

To reduce the risk of infection from intravenous solutions due to contamination, appropriate handling, storage, and administration are key. Solutions with added substances (e.g. electrolytes, medication, dextrose) should be used within 24 hours. Lipid-containing fluids have a higher risk of contamination and bags should be discarded at the end of the day, regardless of whether or not they are empty. See Chapter: Blood Donation for more information on management of blood and blood products from donors.

Catheter-associated urinary tract infections (UTIs)

Catheter-associated UTIs are common HAIs in small animal clinics (Stull & Weese 2015). The risk of a UTI increase every day a patient is catheterized with a urinary catheter (Smee 2013). The means by which the urinary tract becomes infected and the origin of the bacteria are highly variable. Bacteria may be endogenous or introduced during catheter placement. Retrograde urine flow allows exogenous and endogenous bacteria to move up the catheter into the bladder. Biofilms may also result in UTIs, and are associated with lack of response to antimicrobial therapy.

Using aseptic technique and managing the catheter appropriately reduce the risk of UTI development:

- Ensure that the type of catheterization (intermittent vs. indwelling), catheter selection, placement, maintenance, changing and timing of removal are tailored to the patient and situation. The patient should be reassessed regularly to verify whether ongoing catheterization is required.
- Use only sterile urinary catheters (either new or resterilized).
- Gently but thoroughly clean the area around the urethra prior to catheter placement. Clip long hair if necessary. Aseptic skin preparation should be used if placing a percutaneous urinary catheter. Avoid causing excessive irritation to the area which could increase the risk of bacterial colonization and infection.
- Use sterile gloves and lubricant for placement. Perform hand hygiene before gloving and after glove removal.
- Follow appropriate antimicrobial therapy guidelines such as those available from ISCAID (Weese 2019) (also see Chapter: Antimicrobial Stewardship).
- Keep the urine collection bag below the level of the patient at all times to prevent backflow of urine.
- Never leave the catheter or tubing line open to the outside environment. Always keep it connected to a urine collection bag.
- Do not routinely culture urine from patients with indwelling urinary catheters (Weese 2019). Never collect urine for culture from the collection bag.

Surgical site infections (SSIs)

See Chapter: Surgery.

Other sources of HAIs

Ventilator-associated pneumonia (VAP)

Although VAP is relatively common in human intensive care units, little is known about VAP in animals due to the relatively low use of long-term mechanical ventilation outside of procedures done under general anesthesia. General infection control practices will reduce the risk of VAP, in addition to proper equipment and filter maintenance. Veterinary specific guidelines for VAP prevention are not available, but some recommendations from human healthcare guidelines may apply (CPSI 2012) (also see Chapter: Cleaning, Disinfection, Sterilization).

Endoscope-associated infections

Proper cleaning and disinfection of endoscopes are the cornerstones of endoscope-associated infection prevention (see Chapter: Cleaning, Disinfection, Sterilization). Endoscope culture is indicated when endoscopes are implicated as a source of potential infection during a disease outbreak investigation, along with careful review of endoscope cleaning and disinfection protocols and practices. In practices where multiple endoscopes are available, the specific endoscope used for each procedure should be documented to allow for accurate contact tracing.

Contamination of enteral feeding solutions

Contamination of liquid diets used for enteral (tube) feeding typically occurs during mixing, or during administration, particularly when they need to be given very slowly over a prolonged period, or continuously. To reduce the risk of contamination:

- Use products that do not require any mixing or additives.
- Always follow the manufacturer's recommendations for storage.
- Thoroughly rinse and clean bags and lines in hot water between uses for the same patient. Discard or thoroughly scrub and then disinfect (e.g. with an accelerated hydrogen peroxide product or a 1:50 concentration of bleach) prior to use on a different patient.
- Maximum hang time for enteral solutions should be 4-6 hours. Label bags with the time they were opened, and discard them after the maximum time.

Management of specific infectious syndromes

Implementation of basic infection control measures is important for handling all patients, but especially those with active infections that are likely to be shedding much higher numbers of transmissible pathogens. Animals with confirmed or suspected multidrug-resistant (MDR) infections or at risk of shedding an infectious gastrointestinal or respiratory pathogen should be treated with the same enhanced infection control precautions. These precautions should include front office triage, contact precautions and personal protective equipment (PPE), housing in isolation and other patient handling protocols, and cleaning and disinfection of in-contact equipment and environmental surfaces (see the beginning of this chapter for links to the corresponding chapters on these topics).

Multidrug-resistant (MDR) infections

Multidrug-resistant organisms (MDROs) are becoming commonplace in both human and veterinary medicine. As such, it is important to have practices in place to manage patients with suspected or known MDR infections.

Infections with MDR bacteria, such as *Staphylococcus aureus* (MRSA) and *S. pseudintermedius* (MRSP), can be managed with relatively simple, cost effective procedures that are applicable for a very broad range of other pathogens as well. Hand hygiene, environmental cleaning and disinfection, and sound antimicrobial stewardship are the pillars of prevention and control in these cases. Other infection control practices and aseptic techniques will help prevent pathogen spread and improve management of patients with known or suspected infections. When handling a patient with an MDRO:

- Keep broken skin covered to help prevent opportunistic infection with the MDRO.
- Pay particular attention to hand hygiene, as hands of personnel are one of the most common means of spread in the hospital setting.
- If the MDRO is associated with a urinary tract infection, label urine samples as infectious and handle accordingly.
- Carefully manage wounds (see wound care and bandages section in Chapter: Patient Care and Handling).

There is no clear evidence on how long an animal with an MDR infection should continue to be handled with contact precautions following diagnosis. At a minimum, the animal should be considered infectious to others until the clinical infection is resolved (e.g. until the animal is no longer being seen for rechecks of the same issue). Some pathogens such as MRSP may be carried for prolonged periods of time at other body sights even after clinical resolution of the original infection.

Ringworm and other non-bacterial skin infections

Non-bacterial skin infections requiring additional considerations include primarily dermatophytosis (ringworm) and sarcoptic mange, both of which are easily spread between patients. *Sarcoptes* mites are fairly species-specific and at worst would only transiently infest a person or the clinic environment. However, certain dermatophytes can easily be transmitted to people as well as other patients, and these fungi tend to survive very well in the environment within dust, dander and hair shed by the infected animal. Additional precautions to consider when dealing with such cases include:

- Keep the patient in its own run/kennel as much as possible. For dogs that must be taken out for elimination activities, avoid any direct contact with any other patients.
  - After an infected dog is discharged, soak the leash in bleach solution (1:50) for 10-30 mins or an accelerated hydrogen peroxide (AHP) solution (as per label instructions), or autoclave if possible.
- Depending on the size/type of animal, appropriate PPE may need to include long pants or a longer gown to protect the legs, which must be removed and laundered or disposed each time after handling the patient.
- Avoid dry dusting, as this can spread contaminated hair and dander within a room. If possible, wet surfaces first and wipe with a damp towel/cloth.
- Use a disposable mop head to clean hard floors.
- If vacuuming is necessary, ensure the vacuum is equipped with a HEPA filter, and dispose of the filter immediately once vacuuming is done. Without such a filter, vacuuming may result in further spread of contaminated hair and dander within the room.

- All surfaces and reusable items that come into contact with the infected animal must be cleaned with hot, soapy water, followed by an effective disinfectant:
  - household bleach (1:10 to 1:100 solution in water)
  - lime sulfur (1:33 solution)
  - enilconazole (0.2% solution)
  - AHP product
- Avoid keeping infected patients in rooms with a cold air return.
- Fabric items (e.g. drapes, carpet, cushions) that cannot be effectively laundered may need to be discarded to eliminate dermatophytes if infections continue to occur after all other precautions have been taken.

An infosheet for veterinarians on ringworm in companion animals is available from the Worms & Germs Blog (see References).

Leptospirosis

In the case of animals infected with *Leptospira* spp. special attention should be paid to items that have come into contact with the animal's urine. Immunocompromised staff in particular should avoid any contact with leptospirosis patients if at all possible. Of particular note:

- Cover broken skin and wear a face mask/goggles if there is a splash risk.
- Do NOT pressure wash kennels.
- Keep surfaces dry as much as possible.
- Normal laundering inactivates leptospires; however, personnel must wear proper PPE (i.e. gloves and designated lab coat) when handling bedding.
- After cleaning, disinfect areas where dogs have urinated with and AHP or other effective product according to label directions, or a 1:10 bleach solution.
- Label urine samples as infectious and handle them accordingly.
- House patients on ground level.
- Allow urination to occur on cleanable surfaces that are designated and restricted.
  - Be aware of the potential for urine to contaminate the animal's coat during elimination, especially if the animal urinates on a hard surface.
- Collected urine must be inactivated before disposal using a 1:1 ratio of an appropriately diluted disinfectant solution such as an AHP, quaternary ammonium compound (QAC), 1:10 bleach, or iodine.
- Critically ill leptospirosis patients can be effectively managed outside of isolation if necessary, but then procedural isolation, signage and clear communication with all clinic staff regarding appropriate protocols become even more crucial.
- 48 hours after the commencement of appropriate antimicrobial therapy, live/infectious leptospires are thought to no longer shed in the urine (Sykes 2010), and isolation precautions may no longer be necessary. Before contact precautions are relaxed, ideally the animal should be bathed and dried thoroughly to ensure any urine contamination of the coat has been eliminated.

An infographic on in-clinic management of leptospirosis patients is available from the Ontario Animal Health Network (see References).

Gastrointestinal infections

Diarrhea is usually a readily-apparent clinical sign in animals, however its cause is often unknown. Some causes may be zoonotic, such as *Salmonella* and *Campylobacter*. Certain populations may be at greater risk of shedding certain pathogens, such as puppies and kittens *(Campylobacter),* dogs fed animal-based food items (e.g. raw meat, pig ear treats) or with exposure to livestock *(Salmonella, E. coli, Listeria),* and reptiles *(Salmonella).*

Although the zoonotic transmission of diarrhea-causing pathogens is poorly understood, *Salmonella* and *C. difficile* have been identified in veterinary staff (Marks 2011). Soap and water hand hygiene may be preferrable in some of these cases as bacterial spores (e.g. *Clostridium* spp.) are resistant to alcohol, as are some non-enveloped viruses (e.g. parvovirus) that can

also cause diarrhea, but are not zoonotic. Handwashing with soap and water is certainly necessary if the hands are grossly contaminated with diarrhea, as the organic material inhibits the antimicrobial action of alcohol-based hand sanitizers.

Patients at increased risk of shedding enteropathogens (e.g. those who are diarrheic or recently diarrheic) should be taken to a designated location for defecation, away from other patients. The area should be disinfected, if possible. Dogs should remain on leash and only defecation and urination should occur in this area, with feces cleaned up immediately. Litter boxes for cats should be regularly cleaned and disinfected to decrease the environmental pathogen load in the patient's housing area.

Should a patient pass diarrhea unexpectedly in any other area of the hospital:

- Remove feces immediately and clean the area using disposable towels.
- Liberally apply disinfectant to the area:
  - Bleach (1:10 solution) or AHPs (dilutes as per label instruction) are effective. Both C. *difficile* and C. *perfringens* spores are susceptible to these solutions.
- Leave the disinfectant in contact for the required contact time on the label (typically 10-15 minutes). Ensure all surfaces remain wet for the entire duration.
- Rinse the area if necessary to eliminate residual disinfectant, and dry all surfaces completely (either air dry or by means of towels/fans if area must dry faster).
- Treat all waste/soiled linens (e.g. disposable and reusable towels) as infectious and handle accordingly.

Respiratory infections

Control of respiratory disease outbreaks in veterinary clinics is dependent on prompt recognition of the potential for an infectious pathogen, the ability to effectively separate (procedurally and/or physically) affected and unaffected animals, and strict adherence to general principles of infection control.

Routine use of mask and eye protection is not required, but should be considered in situations where someone's face will be in close proximity to a potentially infected animal, especially if the animal is coughing. Routine disinfectants, used properly, will inactivate the vast majority of respiratory pathogens. Enveloped viruses such as influenza A virus are particularly fragile and susceptible to disinfectants. Ensuring all contaminated surfaces and equipment are addressed and thoroughly cleaned before applying disinfectant is important. Veterinary personnel should pay particularly close attention to hand hygiene. Alcohol-based hand sanitizers are effective against the vast majority of respiratory pathogens, or hands can be washed with soap and water.

An infosheet for veterinarians on H3N2 influenza in dogs is available from the Worms & Germs Blog (see References).

References

British Small Animal Veterinary Association. BSAVA practice guidelines: Reducing the risk from MRSA and MRSP. Available at: https://www.bsava.com/Portals/0/resources/documents/BSAVA_MRSA_Guidelines_0711.pdf. Accessed Dec-2018.

Canadian Patient Safety Institute (CPSI). Safer Healthcare Now! Prevent ventilator associated pneumonia: Getting started kit. 2012. Available at: https://www.patientsafetyinstitute.ca/en/toolsResources/Documents/Interventions/Ventilator-Associated%20Pneumonia/VAP%20Getting%20Started%20Kit.pdf#search=VAP. Accessed Dec-2018.

Davis H, et al. 2013 AAHA/AAFP fluid therapy guidelines for dogs and cats. J Am Anim Hosp Assoc. 2013;49:149-159.

Gober M, McCloskey R. Canine infectious respiratory disease (CIRD): Management of outbreak situations. Zoetis Technical Bulletin. 2013. Available at: https://www.zoetisus.com/products/dogs/bronchicine/pdf/cird_technical_bulletin.pdf. Accessed Dec-2018.

Goldstein RE. Canine leptospirosis. Vet Clin North Am Small Anim Pract. 2010;40(6):1091-101. PubMed PMID 20933138.
Hillier A, et al. Guidelines for the diagnosis and antimicrobial therapy of canine superficial bacterial folliculitis (Antimicrobial Guidelines Working Group of the International Society for Companion Animal Infectious Diseases). Vet Dermatol. 2014;25(3):163-e43. PubMed PMID: 24720433.

Lappin MR, et al. Antimicrobial use guidelines for treatment of respiratory tract disease in dogs and cats: Antimicrobial Guidelines Working Group of the International Society for Companion Animal Infectious Diseases. J Vet Intern Med. 2017;31(2):279-294. PubMed PMID: 28185306.

Marks SL, et al. Enteropathogenic bacteria in dogs and cats: Diagnosis, epidemiology, treatment, and control. J Vet Intern Med. 2011;25:1195-1208.

Mathews KA, et al. A prospective study of intravenous catheter contamination. J Vet Emerg Crit Care. 1996;6(1):33-43.

Ontario Veterinary College (OVC). Ontario Veterinary College Health Sciences Centre: Infection Control Manual. 2011. Available at: https://ovc.uoguelph.ca/doc/InfectionControlManual_Aug_2011_V2.pdf. Accessed Dec-2018.

Schuller S, et al. European consensus statement on leptospirosis in dogs and cats. J Small Anim Pract. 2015;56(3):159-79. PubMed PMID 25754092.

Smee N, et al. UTIs in small animal patients: Part 2: Diagnosis, treatment, and complications. J Am Anim Hosp Assoc. 2013;49:83–94. PubMed PMID 23325594.

Stull JW, Weese JS. Hospital-associated infections in small animal practice. Vet Clin North Am Small An Pract. 2015;45:217-233.

Sykes JE, et al. 2010 ACVIM small animal consensus statement on leptospirosis: Diagnosis, epidemiology, treatment, and prevention. J Vet Intern Med. 2011;25:1-13. PubMed PMID 21155890.

O'Grady NP, et al. Guidelines for the prevention of intravascular catheter-related infections. Clin Infect Dis. 2011;52(9):e162-93. PubMed PMID: 21460264.

Ontario Animal Health Network (OAHN). Brush up on managing lepto patients (infographic). 2016. Available at: http://oahn.ca/resources/brush-up-on-managing-lepto-patients-a-leptospirosis-infographic/. Accessed Dec-2018.

Weese JS, et al. International Society for Companion Animal Infectious Diseases (ISCAID) guidelines for the diagnosis and management of bacterial urinary tract infections in dogs and cats. Vet J. 2019;247:8-25. PubMed PMID: 30971357.

Worms & Germs Blog. H3N2 canine influenza for veterinarians (infosheet). University of Guelph. 2018. Available at: https://www.wormsandgermsblog.com/files/2018/01/H3N2-CIV-Infosheet-V1.pdf. Accessed Dec-2018.

Worms & Germs Blog. Ringworm for vets (infosheet). University of Guelph. 2008. Available at: https://www.wormsandgermsblog.com/files/2008/04/M2-Ringworm-DVM.pdf. Accessed Dec-2018.

Cleaning, Disinfection, Sterilization

Cleaning and disinfection are two separate tasks. **Cleaning** involves the removal of visible organic matter and scrubbing with soap or detergent, whereas **disinfection** involves the application of a chemical or other procedure in order to kill the majority of microbes that cannot be adequately removed by cleaning. **Sterilization** is the elimination of all viable microbes, including those that are not killed by disinfection. Cleaning is essential because the survival time of many infectious agents outside the host is prolonged by the presence of organic matter, and organic matter also decreases the effectiveness of disinfection and sterilization. Depending on the level of disinfection used, disinfection kills or prevents the growth of many or most pathogens.

Equipment should be cleaned and disinfected according to its intended use, the manufacturer's recommendations, and practice policy. Equipment must be cleaned before disinfection or sterilization. Surfaces where animals are housed, examined, or treated should be made of non-porous, sealed, easy-to-clean materials to facilitate cleaning and disinfection and minimize pathogen transmission.

Personnel whose duties include cleaning and disinfection or sterilization of equipment and different hospital areas should be trained on how to safely handle and use the products available in the clinic for these procedures. In Canada, Material Safety Data Sheets (MSDS) must be readily accessible for all applicable chemical products.

Cleaning

Cleaning entails the removal of all visible organic matter of any kind (e.g. feces, urine, blood, food, dirt etc.) from a surface using soap, detergent, or other such product, or other physical means (e.g. ultrasonic cleaner). It is especially important in the presence of pathogens that are resistant to most disinfectants (e.g. *Cryptosporidium* spp.). The mechanical action of cleaning physically removes these organisms from a surface. It is necessary for both environmental and equipment surfaces (see Sterilization section for additional information on cleaning instruments and equipment prior to sterilization). Cleaning must always be done before a disinfectant is used or before sterilization is performed.

Cleaning procedures

When using a cleaning product:

- Ensure the area is well ventilated.
- Bear in mind that during cleaning, it is the mechanical action of scrubbing and the surfactant properties of the product that are important, not the product's antimicrobial activity.
- Wear gloves during the procedure, and wash hands once complete. Other personal protective equipment, such as eye protection or respiratory protection, may be required depending on the situation.
- Ensure any residue from chemical cleaning products is rinsed off or otherwise removed when done.
- Allow all surfaces to dry completely once complete.

Recommended cleaning procedures for common environmental surfaces are shown in Table 1.

Removing loose, dry debris from surfaces:

- Avoid generating airborne dust that may contain pathogens by:
  - using a vacuum cleaner equipped with a HEPA filter
- The filter helps to prevent aerosolization of pathogens such as ringworm. For this reason, vacuums without HEPA filters should not be used for cleaning in patient areas.
  - lightly spraying surfaces with water prior to wiping or sweeping
  - using an electrostatic wipe (e.g. Swiffer™ cloth)
  - using a wet mop

- Exposure to aerosols generated by brushes during cleaning can be minimized by taking certain precautions, such as wearing a face mask and containing spatter if the brush or surface is damp. A surgical nose-and-mouth mask will provide some protection against droplet spatter. For finer particles and dry dust that can become suspended in the air, a properly-fitted N95 respirator is needed for protection (see respiratory protection section in Chapter: Personal Protective Equipment).

Removing sticky, wet or dried-on organic material from surfaces:

- Use a detergent or soap and a brush or cloth, as necessary.
- Avoid the use of pressure washers, particularly those that produce more than 120 psi. This amount of pressure may cause aerosolization of pathogens, and may even damage surfaces, thus making them harder to clean and disinfect over time. A home garden hose sprayer usually produces less than 120 psi of pressure, and would therefore be relatively safe to use in a small animal kennel area.

Disinfection

Disinfection can only be maximally effective if it is preceded by thorough cleaning. Some pathogens (e.g. clostridial spores) are highly resistant to disinfection, therefore cleaning in these cases is particularly crucial in order to mechanically remove the organisms.

Disinfection procedures

When using a disinfectant (adapted from PHO 2018):

- Ensure all areas are well ventilated.
- Wear gloves when handling disinfectants. Note that latex gloves will lose their integrity when exposed to many chemicals. For small jobs, use disposable nitrile gloves instead. For large jobs, heavier rubber gloves (e.g. common dishwashing gloves) can be used, but reusable gloves of this type must themselves also be disinfected at the end of each task.
- Wear eye protection in case of splashes.
- Always apply the selected disinfectant according to the product label, with particular attention to:
  - appropriate dilution
  - required contact time
- If patients or personnel may have direct skin contact with the surface, if the surface is used for food preparation, or if the disinfectant used may damage a particular surface, the disinfectant may need to be rinsed off with clean water after an appropriate amount of time has elapsed.
- After disinfection, allow all surfaces to dry completely.

It is also important to reduce the risk of contamination of the disinfection equipment and containers. Proper dilution of products, frequent refreshing of solutions, use of clean cloths and wipes, and periodic cleaning and disinfection of the equipment and containers themselves (including complete emptying and allowing to dry) will help achieve this goal. Depending on the product, test strips are available that can be used to verify ongoing activity of the diluted (i.e. in-use) disinfectant if stored or maintained in basins for a long duration. Also see References for an online bleach dilution calculator (PHO).

Environmental cleaning and disinfection

Environmental surfaces that are not in direct contact with patients are generally low risk in terms of contributing to pathogen transmission. Routine cleaning with or without disinfection are adequate for these surfaces (e.g. walls, chairs, monitors, cabinets) in most cases. See Table 3 for recommendations regarding cleaning and disinfecting such surfaces. Compared to human healthcare facilities, particular attention needs to be paid to the cleaning and disinfection of floors, as patients and personnel often have very close contact with the floor.

Cleaning must always be done before a disinfectant is used or before sterilization is performed.

TABLE 3. Recommended cleaning and disinfection procedures for common environmental surfaces (also see disinfectant selection section).

Surface/Object	Procedures	Special Considerations
Horizontal surfaces with low patient contact (e.g. front desk, records area)	• Clean regularly with detergent, e.g. bi-weekly • Clean and disinfect promptly if visibly soiled with feces, urine or body fluids	Electrostatic wipes (e.g. Swiffer™ cloths) can be used to remove loose fur and dust
Horizontal surfaces with high patient contact (e.g. exam tables, scale, kennels)	• Clean and disinfect between all patients • Provide enhanced disinfection after contact with high-risk patients (e.g. diarrheic). A broader-spectrum disinfectant (e.g. bleach, oxidizing agent) should be used if narrower-spectrum disinfectants are used routinely (see disinfectant selection section)	
Vertical surfaces (e.g. walls, doors, windows including blinds/ curtains)	• Clean regularly with a detergent, e.g. monthly • Clean and disinfect promptly if visibly soiled with feces, urine or body fluids	
Hard floors (e.g. tile, wood, sealed cement)	• Clean daily with a detergent and disinfect regularly, e.g. weekly • Clean and disinfect after potentially infectious patients • Clean and disinfect promptly if visibly soiled with feces, urine or body fluids	
Carpets/upholstery	• Vacuum regularly, e.g. monthly • Use a vacuum equipped with a HEPA filter, especially if there may have been contact with an animal shedding an infectious pathogen (e.g. ringworm) • Shampoo or steam clean if necessary to remove persistent dirt and stains	Cleaning is especially important for these surfaces as they are difficult or impossible to disinfect
Food and water bowls	• Cleaning alone (with regular dish soap) is adequate for food and water bowls from non-infectious patients • Clean and disinfect after potentially infectious patients, and do not clean with bowls from other patients, or use disposable bowls	Use a disinfectant approved for use on food surfaces
Toys and miscellaneous items	• Clean and disinfect between patients, or discard if not amenable to proper cleaning and disinfection (e.g. cloth toys)	Wear gloves when handling items from patients with known or suspected zoonotic pathogens or any items that are visibly soiled
Litter boxes	• Clean out at least daily • Completely empty and disinfect between patients	Ideally, litter boxes should not be handled by pregnant women, however daily cleaning & disinfection reduces the risk of contact with the infectious stage of certain parasites

Footbaths and footmats

Data regarding the need for and efficacy of footbaths and footmats are very limited, and there is essentially no information related to small animal clinics specifically. It has been shown that footbaths can reduce bacterial contamination of footwear in large animal clinic settings (Hornig 2016). Although other sources of contamination have been shown to be more significant in infection transmission, footwear and floor surfaces cannot be overlooked in an infection control program in a small animal clinic, because patients so often have extensive direct contact with the floor. In addition, footbaths or footmats can be a useful visual indicator of an area of increased infectious disease risk.

Footbaths are shallow containers filled with a disinfectant solution. Footmats are spongy mats covered with a durable, easy-to-clean material that can be saturated with disinfectant. Footmats can increase compliance because they are easier to use, but they are more expensive and more difficult to maintain than footbaths. Footbath or footmat use is almost invariably accompanied by spillage of disinfectant solution; this can create a slipping hazard on smooth floor surfaces, which are typically present in small animal clinics. Slipping risks can be reduced with the use of a "walk of"' mat to provide traction. Some disinfectants can also damage floor surfaces with prolonged contact.

Footbaths or footmats should be considered when personnel will be walking on a surface that could potentially be more contaminated than the rest of the floor in general, and where spread of this contamination might pose a risk to patients or personnel. The most likely area where footbaths or footmats could be useful is at the exit of an animal housing area (e.g. dog run) that contains a potentially infectious case, and where clinic personnel will be walking in and out of the potentially contaminated area. The need for routine use of footbaths or footmats in isolation areas where animals are confined in cages is questionable. If used, select a disinfectant that is effective against the specific pathogen(s) of concern, stable in solution, and effective with a relatively short contact time (see Tables 4 and 5). Oxidizing agents such as accelerated/stabilized hydrogen peroxide and peroxygen disinfectants are ideal. The solution should be changed daily, or sooner if gross contamination of the bath/mat occurs. Maintaining proper concentration of active disinfectants in footbaths and footmats is essential for proper performance.

Maintaining proper concentration of active disinfectants in footbaths and footmats is essential for proper performance.

Equipment disinfection

Single-use equipment (e.g. hypodermic needles) should not be re-sterilized or disinfected for re-use. Such items should be properly disposed of immediately after initial use. In veterinary medicine, although it is not considered best-practice, some equipment that is classified as single-use in human healthcare is reused because the cost of some items makes it impractical to discard them after a single use (see disinfection of anesthetic equipment section in Chapter: Surgery). There is little to no objective information on how to disinfect or sterilize such equipment, and how often this can be done without compromising the integrity of the item. The level of disinfection required should be evaluated as for multi-use equipment (below). Items should be carefully inspected prior to each use and replaced if there is evidence of damage that may impair the function of the equipment or subsequent cleaning and disinfection. In some areas, licensed third party companies may exist that can provide reprocessing services for some equipment.

Multi-use equipment must be properly cleaned and disinfected between each patient. There are three categories of multi-use equipment used on patients: **critical, semi-critical** and **non-critical.** Each category defines how instruments must be cleaned and disinfected to prevent transmission of infectious agents. In human healthcare, these categories are defined as per Table 4.

TABLE 4. Spaulding's classification of medical equipment / devices and required levels of processing and reprocessing (Spaulding 1970).

Classification	Definition	Level of processing / reprocessing
Critical equipment/device (e.g. surgical instruments)	Equipment/device that enters sterile tissues, including the vascular system	Cleaning followed by sterilization
Semi-critical equipment/ device (e.g. endoscopes, thermometers)	Equipment/device that comes in contact with non-intact skin or mucous membranes but does not penetrate them	Cleaning followed by high level disinfection (as a minimum), sterilization is preferred
Non-critical equipment/device (e.g. stethoscope)	Equipment/device that touches only intact skin and not mucous membranes, or does not directly touch the patient	Cleaning followed by low level disinfection, in some cases, cleaning alone is acceptable

See Tables 5 (below), 6 and 7 for selection of disinfectants.

The CDC defines **high level disinfection** as use of a chemical agent capable of killing all vegetative microorganisms, but not all bacterial spores. These agents can be used as chemical sterilants, but should not be used for general environmental cleaning. **Low level disinfection**, on the other hand, is use of an agent capable of killing vegetative bacteria, some viruses and fungi, but no bacterial spores. These products may lack a tuberculocidal claim as well. They are ideal for environmental cleaning and disinfection. Table 5 below lists common disinfectants used for the different levels of disinfection. Additional information is available from various online resources (PHO 2018, Rutala 2008).

In veterinary medicine, exceptions to the level of processing required are typically made for some pieces of semi-critical equipment that come in contact with tissues or mucous membranes which are normally considered non-sterile, such as those of the upper respiratory or gastrointestinal tracts. If a transmissible infectious disease is not suspected in the patient, and the subsequent patient is not significantly immunocompromised, thorough cleaning and low level disinfection is likely adequate in these cases. However, if an infectious disease is suspected or the subsequent patient is immunocompromised, then cleaning and high level disinfection or sterilization are recommended in order to prevent disease transmission. For example, a rectal thermometer should undergo cleaning and low level disinfection between every patient, but if used on a diarrheic animal it should undergo high level disinfection, be discarded and replaced, or disposable protective covers should be used followed by disinfection.

TABLE 5. Common chemical disinfectants (Modified from PHO 2018, Rutala & Weber 2016)

	Low Level Disinfection	High Level Disinfection	Sterilization
Equipment	Non critical equipment	Semi-critical equipment	Critical equipment
Chemical	60-95% Alcohol 0.5% AHP (1-5min) 1:100 bleach (10min) QACs 3% HP	1% HP + 0.08% PA (25min) 7.35% HP + 0.23% PA (15min) >2% glut (20-90min) 2% HP (8min) 0.55% OPA (12min) 2% AHP (5-8mins) 1:50 bleach (10min)	Vapourized HP Ozone HP/ozone >2% glut (10 hours) 0.2% PA (12mins at 50-56C) 6-25% HP (6hours) 2% AHP (6 hours) 7% AHP (20mins) 1:10 bleach (10min)

Contact times and concentrations are guidelines only. Always follow the manufacturer's instructions for the specific product being used.
glut – glutaraldehyde, **HP** – hydrogen peroxide, **PA** – peracetic acid, **OPA** - Ortho-phthalaldehyde, **AHP** – accelerated hydrogen peroxide, **QAC** – quaternary ammonium compound

The following section outlines basic cleaning and disinfection recommendations for select equipment. Detailed instructions for these and other devices should be available from the manufacturer.

Endoscopes: Proper cleaning and maintenance of endoscopes are important to prolonging the useful life of the instrument, but cleaning and disinfection are also important in terms of infectious disease control. Endoscopes are semi-critical equipment, and as such require high-level disinfection when used in humans. In veterinary medicine, high-level disinfection is required prior to use in relatively sterile areas (e.g. urinary tract), and should be considered best-practice for use in in non-sterile areas as well (e.g. gastrointestinal tract, upper respiratory tract), even though thorough low-level disinfection is often used for the latter. Manufacturers typically provide detailed reprocessing (cleaning and disinfection) instructions for their instruments, which should be readily available as a reference for staff members responsible for the care of endoscopes. If the endoscope was purchased second hand and the reprocessing instructions were not provided, it is important to contact the manufacturer to obtain a copy. Some general guidelines regarding endoscope maintenance are as follows (adapted from Rutala 2008):

1. **Clean: Endoscopes (and associated equipment) must be meticulously cleaned immediately after every use.** This involves first removing visible organic matter to prevent debris from drying onto surfaces, as this can make the debris considerably harder to remove. Endoscopes typically have several moving or detachable parts and small channels in which moisture, debris and discharge can become trapped. After cleaning the visible debris from the scope, it should be disassembled and cleaned with water and a detergent or enzymatic cleaner. Leak testing should be done prior to immersion. The scope should be dried before proceeding to disinfection.
   - **All instrument and suction channels must be thoroughly cleaned after each use**, even if the channels were not used during the procedure. Failure to clean these channels is a common error which can result in accumulation of debris, bacteria and biofilms within the instrument. Not only does this pose a risk of disease transmission to subsequent patients, but it can also confound sample collection and culture.
2. **Disinfect:** All channels of the endoscope must be disinfected using a **low level disinfectant, at a minimum**. High level disinfection is required prior to procedures involving sterile areas, and should be considered best-practice prior to any use. Immersion of the endoscope in the solution and perfusion through the channels to remove air pockets ensures all surfaces come into contact with the disinfectant. Consult the manufacturer's instructions regarding what methods can be safely used for any particular endoscope. If a chemical sterilant is used, **a timer should be used to measure the exact contact time** — too short a time may result in an inadequate microbial killing, while too long a time may result in damage to the instrument.
3. **Rinse:** Failure to rinse off detergents or disinfectants can lead to significant irritation of the tissues of the next patient. Rinse using filtered water or high quality potable tap water.
4. **Dry: Rinse all channels with alcohol,** followed by forced dry air prior to storage (e.g. syringe with room air, assuming medical-grade air is not available).
5. **Store:** Hang endoscopes vertically to prevent recontamination and promote drying.

Clippers: Use of good-quality clippers and maintenance of clipper blades are of great importance, as their use can otherwise result in skin trauma, with subsequent risk for infection or transmission of opportunistic pathogens between patients.

Following routine use of clippers on areas of unbroken skin and non-infectious animals, **basic cleaning with a stiff brush** to remove visible dirt and hair from the blade is adequate. More thorough cleaning and disinfection of the blade, as described below, should be done periodically as well, depending on how often the clippers are used.

Thoroughly clean and disinfect clippers after every use on an animal with a potentially transmissible infection (e.g. an animal with diarrhea), on any area where the skin or hair is significantly contaminated with feces, urine, blood or other body fluids, and before and after use on an area where the skin is broken (especially if there is evidence of skin infection).

- Use a stiff brush to remove visible dirt and hair from the blade, and a soapy, wet cloth to remove any visible debris from the body of the clippers. Blades with broken teeth or that have become dull over time should be discarded and replaced.

- The clipper blade can then be sterilized using a chemical sterilant (e.g. glutaraldehyde) or by autoclaving.
- The body of the clippers can be sterilized using hydrogen peroxide vapour or ethylene oxide (if available). Otherwise, after removing all visible debris, use a disinfectant wipe or cloth wetted with a routine disinfectant solution to thoroughly wipe down the body of the clippers, paying particular attention to the small crevices of the device and allowing for adequate contact time with the disinfectant. Refer to the clipper's instruction manual to determine what degree of contact with liquid the clippers can safely withstand.

Thermometers: By virtue of their contact with the rectum, rectal thermometers are at high risk of becoming contaminated with a variety of pathogens. Measures that can be taken to reduce the risk of pathogen transmission include dedicating thermometers to individual patients, use of disposable thermometer covers, and use of proper disinfection practices. The most practical approach is to clean the thermometer to remove gross contamination, and then wipe with a disinfectant, such as an accelerated hydrogen peroxide, that is able to kill non-enveloped viruses and bacterial spores. This is unlikely to completely eliminate all pathogens but may adequately reduce contamination to reduce disease risk. Special attention should be paid to thermometers used with infectious cases. Ideally these thermometers should be discharged with the patient or discarded, however this may be impractical in many facilities. High level disinfection after such cases should be used to reduce the risk that harmful pathogens remain present on the device.

Ophthalmoscopes and other hand-held instruments: Handheld equipment such as ophthalmoscopes and otoscopes are frequently touched but may be infrequently cleaned. While their role in pathogen transmission is likely limited, routine cleaning and disinfection of these instruments (like any common hand contact surface) is indicated. There are no objective guidelines for frequency of cleaning and disinfection, but incorporating these items into a regular (e.g. weekly) schedule is logical, with additional cleaning and disinfection done on an as-needed basis after use on any potentially infectious patient. Removal of visible debris followed by wiping with disinfectant is practical for other equipment. Some items may be more difficult to disinfect based on their design (e.g. many crevices) or surface materials. These factors, and material compatibility, must be considered when selecting an appropriate disinfectant. Single-use disposable covers should be used for instruments such as tonopens when available/applicable.

Otoscope cones: While manufactured as single-use items, otoscope cones are commonly re-used in veterinary practices. Because of the contact with the ear canal, often in patients with otitis, contamination of otoscope cones is common. Cleaning must occur prior to disinfection to remove all organic material (e.g. earwax, exudates). Disinfection can be accomplished by soaking in 2% chlorhexidine solution. Chlorhexidine is primarily used as an antiseptic, but has been recommended for use with otoscope cones due to the amount of contact with the relatively bare and often sensitive skin of the ear canal. Use of a quaternary ammonium compound (QAC) or accelerated hydrogen peroxide (AHP) product would also be a reasonable choice. Ensure adequate concentration and contact times are used as per the manufacturer guidelines. Containers of standing solution/baths must also be cleaned and refreshed regularly. Regardless of the disinfectant used, cones should still be rinsed with water and dried prior to use to prevent tissue irritation from the disinfectant.

Ultrasound units and other patient-side equipment: Patient-side medical equipment can easily become contaminated with pathogens from direct contact with patients, other contaminated items, and hands of veterinary personnel. Specific recommendations are difficult to provide because of the variability in equipment surfaces, but the basic principles of cleaning and disinfection still apply:

- **Prevent contamination:** While it is impossible to prevent all contamination, the incidence and degree of contamination can be reduced by limiting direct patient contact with equipment as much as possible, using personal protective equipment (e.g. gloves) as indicated and using barriers (e.g. plastic sleeves) for high-risk (or all) cases. If the patient is harbouring a pathogen of particular concern, consider whether the procedure is truly necessary or can be postponed, particularly if the equipment cannot be reliably disinfected.
- **Clean:** Wiping equipment to remove visible contamination will greatly reduce any pathogen burden.

- **Disinfect:** Specific disinfection procedures will vary with the surface material and characteristics, but wiping with a routine environmental disinfectant is usually practical and likely effective. Consideration must be given to sensitive equipment (e.g. ultrasound probe heads) and manufacturer guidelines should be consulted with respect to compatibility with different disinfectants.

Disinfectant selection

There is no "standard" disinfection program that can be used in all veterinary clinics, as clinic environment, surfaces, caseload, general practices and other factors influence disinfectant choices. Selection of a disinfectant for a particular purpose should take into account the product's spectrum of activity, susceptibility to inactivation by organic matter, potential pathogens in the environment, compatibility with soaps and detergents, toxicity for personnel and animals, contact time required, residual activity, corrosiveness, environmental effects and cost. See Tables 6 and 7 for characteristics and antimicrobial spectrum of different classes of disinfectants. Additional information is available from various online resources (PHO 2018, Rutala 2008).

In order to simplify clinical protocols, ideally one disinfectant should be selected for routine use on most environmental surfaces and non-critical equipment. This product should have a reasonable spectrum of activity and be relatively safe to use for personnel, with minimal requirements for precautions such as ventilation, and a realistic contact time (as per manufacturer guidelines) based on its intended use. A second product should be available for periodic enhanced disinfection when there is suspicion of the possible presence of a pathogen that may not be effectively killed by the disinfectant used routinely (e.g. bacterial spores).

Disinfectant wipes: There is currently little research comparing disinfectant wipes to other disinfection methods. Proper storage is essential to prevent wipes from drying out (thereby reducing their efficacy), and to prevent the introduction of pathogens to the container. The type of disinfectant wipe must also be taken into consideration. Some disinfectant wipes are labeled as one-step, meaning they do not require a pre-cleaning step prior to use. Two-step wipes require the surface to first be cleaned with one wipe, then disinfected with a second wipe. As with any other disinfectant, product labels should be reviewed and the product deemed appropriate for use based on the selection criteria listed above.

Accelerated hydrogen peroxide (AHP): AHP products, also known as improved or enhanced hydrogen peroxide products, have become increasingly popular and more affordable since the early 2000s. This type of disinfectant is highly effective, non-toxic, and non-irritating (when appropriately diluted for use). When combined with surfactants in an acidic solution the hydrogen peroxide's microbicidal activity and cleaning efficacy are enhanced (Rutala 2012). AHPs are considered safe for the environment, people, and animals as they are biodegradable and do not leave any chemical residue on surfaces. AHPs may be used for low or high level disinfection depending on the concentration. AHPs have been shown to be effective with as little as 30-60 seconds of contact time (but as with any product, always follow the manufacturer guidelines for contact time). AHPs are also effective in the presence of limited organic material. However, AHPs can corrode soft metals such as copper, brass, and aluminum, as well as carbon-tipped instruments.

TABLE 6. Characteristics of selected disinfectants (Modified from Linton 1987, Block 2001)

Disinfectant Category	Activity in Presence of Organic Matter	Advantages	Disadvantages	Hazards	Comments
Ethyl alcohol Isopropyl alcohol (60-95% concentration)	Rapidly inactivated	Fast-acting No residue Relatively non-toxic Broad spectrum Inexpensive	Rapid evaporation Coagulates protein, inactivated by organic material May degrade plastic tubing or rubber equipment	Flammable	Not appropriate for environmental disinfection Primarily used as antiseptics
Fomaldehyde Glutaraldehyde	Good	Broad spectrum Relatively non-corrosive	Highly toxic	Irritant Carcinogenic Requires ventilation Coagulates blood, tissues	Used as an aqueous solution or as a gas (fumigation)
Ammonia	Unclear	Some efficacy against coccidial oocysts	Unpleasant odour	Do not mix with bleach Do not add water to lye Irritating Extremely caustic	Not recommended for general use
Chlorhexidine	Moderate	Non-toxic Residual activity increases with more frequent use	Incompatible with anionic detergents	May be associated with skin reactions	Not appropriate for environmental disinfection Primarily used as antiseptic
Hypochlorites (Bleach)	Rapidly inactivated	Broad spectrum, including spores Inexpensive Can be used on food preparation surfaces	Inactivated by cationic soaps/detergents and sunlight Frequent application required	Corrosive Irritant Mixing with other chemicals may produce toxic gas	Used to disinfect clean environmental surfaces Should be stored away from UV and heat
Accelerated hydrogen peroxide (AHP)	Good	Broad spectrum, including spores Environmentally friendly	Breakdown with time	May damage some soft metal surfaces	Excellent choice for environmental disinfection
Peroxygen and organic acid combo	Good	Broad spectrum	Breakdown with time (less readily than AHP)	Corrosive Potentially damaging to surfaces (e.g. concrete)	Good choice for environmental disinfection e.g. Virkon®
Phenols	Good	Broad spectrum Non-corrosive Stable in storage	Toxic to cats Unpleasant odour Incompatible with cationic and nonionic detergents	Irritant Residual film left behind; not appropriate for food surfaces	Some residual activity after drying
Quaternary Ammonium Compounds (QACs)	Moderate	Stable in storage Non-irritating to skin Low toxicity Can be used on food preparation surfaces Effective at high temperatures and pH	Incompatible with anionic detergents	Binding to gauze or cloths may reduce its delivery to target surface	Commonly used primary environmental disinfectant Some residual activity after drying

TABLE 7. Antimicrobial spectrum of selected disinfectants* (Modified from Linton 1987, Block 2001)

Most susceptible

Susceptibility of microorganisms to chemical disinfectants

Most resistant

Agent	Ethyl alcohol Isopropyl alcohol	Formaldehyde	Ammonia	Chlorhexidine Chlorhexidine	Hypochlorite Bleach	AHP, Peroxygen	Phenols	Quaternary Ammonium Compounds
Mycoplasmas	++	++	++	++	++	++	++	+
Gram-positive bacteria	++	++	+	++	++	++	++	++
Gram-negative bacteria	++	++	+	+	++	++	++	+
Pseudomonads	++	++	+	±	++	++	++	±
Enveloped viruses	+	++	+	++	++	++	++	+
Chlamydiae	±	+	+	±	+	+	±	-
Non-enveloped viruses	-	+	±	-	++	+	±**	-
Fungal spores	±	+	+	±	+	±	+	±
Acid-fast bacteria	+	++	+	-	+	±	++	-
Bacterial spores	-	+	±	-	++	+	-	-
Coccidia	-	-	+	-	-	±	+	-

++ Highly effective; + Effective; ± Limited activity; - No activity

Examples of microorganisms from each category:
Mycoplasmas: *Mycoplasma canis, Mycoplasma felis;* **Gram-positive bacteria:** *Staphylococcus* spp, *Streptococcus* spp; **Gram-negative bacteria**: *Bordetella bronchiseptica, Salmonella* spp; **Pseudomonads:** *Pseudomonas aeruginosa;* **Enveloped viruses:** influenza virus, herpesvirus; **Chlamydiae:** *Chlamydophila psittaci;* **Non-enveloped viruses:** feline panleukopenia virus, canine parvovirus; **Fungal spores:** *Blastomyces dermatitidis, Sporothrix schenckii;* **Acid-fast bacteria:** *Mycobacterium avium;* **Bacterial spores:** *Clostridium difficile, Clostridium perfringens;* **Coccidia:** *Cryptosporidium parvum, Isospora* spp, *Toxoplasma gondii.*

*These are intended to be general guidelines for the selection of an appropriate disinfectant. Product labels should always be consulted prior to use to ensure that the efficacy claims are appropriate for the intended purpose.

**In general, phenols are not effective against non-enveloped viruses, but they have been found to be effective against rotaviruses, such as equine rotaviral disease in foals. However, efficacy against small animal parvoviruses has not been demonstrated (Bailey 2013, Stuetzer 2014).

Sterilization

Sterilization involves destruction of all viable microorganisms, including hardy forms such as bacterial spores (Rutala & Weber 1999) and is used for critical items that may come into contact with sterile tissue or the bloodstream (Rutala & Weber 1999, 2009) such as surgical equipment, intravenous catheters and urinary catheters. Sterilization can be achieved through dry heat, steam under pressure, chemical vapours or liquid immersion. Different methods are appropriate for different items and material types. Some of the recommendations below are considered minimum standards in various jurisdictions (e.g. use of autoclave sterilization). Veterinarians should contact their local veterinary regulatory body for details about specific regulations.

Preparation

Items undergoing sterilization must be **clean**. Items that are grossly contaminated (e.g. blood stained) should be considered non-sterile even if they have undergone a sterilization process, as the contamination can physically interfere with the sterilization process.

Specific pre-sterilization cleaning protocols depend on the type of item, manufacturer cleaning recommendations and the degree of contamination. Typically, items are wiped or rinsed to remove gross contamination followed by cleaning with a product to help remove smaller debris, fats, oils and grease. Detergents, enzymatic cleaners and proteolytic products (or combinations thereof) may be used depending on the situation. Detergents help break down oils, fats and grease to facilitate removal and are likely appropriate for most equipment, excluding those coated in organic debris. Enzymatic products are typically used to soak instruments that have dried-on blood or other organic debris, including surgical instruments, and/or instruments with crevices that are hard to clean. Proteolytic products help remove adherent proteins. There is little objective guidance for optimal cleaning practices. After cleaning, items should be rinsed with potable water to remove residual debris and the cleaning agent, since some cleaners may damage surfaces with prolonged contact, and some may be incompatible with chemical disinfectants that are subsequently used.

In veterinary hospitals, most cleaning is done by hand. This is an acceptable approach and it should be performed in a designated clean area (e.g. not in a sink that is likely to be grossly contaminated) and where re-contamination during drying is unlikely. A dedicated sink for reprocessing is ideal. Automated washing machines (e.g. dishwashers, cart washers, specific equipment washers) can be effective and efficient for the purposes of cleaning but must be maintained properly. While some machines have “sanitize cycles”, these should not be considered equivalent to disinfection because of the inability to adequately monitor temperature, contact time and surface coverage.

Instruments should be broken down or disassembled and/or left in the open/unlocked position during sterilization to maximize efficacy of whatever method is used. It is also important not to over pack instruments or attempt to sterilize too many packs in a single load, in excess of the manufacturer’s recommendations.

Sterilization methods

TABLE 8. Sterilization methods*

Approach	Description	Advantages	Disadvantages
Steam/ Autoclave	• Steam sterilization kills through a combination of steam, pressure, temperature and time, with ideal conditions being 100% dry saturated steam with no water (e.g. no mist). • Pressure allows for generation of higher temperatures to more rapidly kill microorganisms • 121ºC for 30 minutes in a gravity displacement autoclave is commonly used • Considered minimum standard for veterinary clinics in some jurisdictions	• Relatively easy • Cost effective • Non-toxic • Dependable	• Not all materials can withstand autoclaving
Immediate-use steam sterilization ("flash" sterilization)	• Process whereby an item is autoclaved in a more rapid manner immediately before use on the patient. • Typical conditions are 132ºC for 3 minutes and 27-28 pounds of pressure with an unwrapped item in a gravity displacement autoclave • Items are generally not wrapped and therefore processed near the patient area where they will be used immediately following sterilization • Most commonly used when an item is dropped during surgery and no sterile replacement is available. • It is also acceptable for items that cannot be packaged, sterilized and stored before use (rare in veterinary clinics) • **Not recommended for routine/general use.**	• Fast	• Less reliable than other methods • Has been associated with SSI and patient burns from hot instruments • Never use for surgical implants
Dry heat	• Uncommonly used • Conditions include 170ºC for 60 min, 160ºC for 120 min or 150ºC for 150 min	• Acceptable for materials that might be damaged by moisture or are impenetrable to moist heat • Non-corrosive	• Long cycle time
Ethylene oxide	• Kills microorganism through alkylation of protein, DNA and RNA • Typical recommended conditions are 450-1200 mg/ml, 29-65ºC, 45-85% and 2-5hr	• Can be used to sterilize items that cannot withstand steam sterilization • Broad spectrum activity against even hardy organisms like mycobacteria, enveloped viruses and bacterial spores	• Cycle time • Cost • Health and safety concerns (chemical is flammable, explosive and toxic) • May be regulatory rules regarding use in some jurisdictions

Approach	Description	Advantages	Disadvantages
Hydrogen peroxide vapour	• Relatively new approach that takes advantage of the broad antimicrobial activity of hydrogen peroxide • Rarely used outside of commercial industry	• No toxic by-products (hydrogen peroxide breaks down to water and oxygen) • Relatively short cycle time • Compatible with a wide range of materials • Can effectively penetrate lumens with small internal diameters or long length	• Expensive
Liquid immersion / cold sterilization	• Involves the use of one of several commercially available chemical sterilants • When used properly, true sterilization can be achieved, but more often used for high-level disinfection • Should be reserved for items that cannot be processed using other methods • The time required to achieve sterilization is typical 3-12 hours (follow manufacturer guidelines) • Items typically need to be rinsed with sterile water or saline (if sterility must be maintained) before being used on patients	• Can be used to sterilize items that cannot withstand steam sterilization (e.g. endoscopes)	• More dependent on thorough cleaning than other sterilization methods • Prone to processing errors (e.g. improper dilution, inadequate contact time, improper pH, failure to ensure no air pockets are present) • Solutions are typically quite toxic and require long contact time and careful management. • Items typically cannot be packaged and stored in a sterile manner after removal from the solution

*always refer to manufacturer recommendations for use of specific equipment and products

"Cold sterile" solutions in veterinary clinics: In some veterinary clinics, disinfectant solutions of various kinds in which a set of instruments is routinely kept are frequently referred to as "cold sterile." Such misuse of this term should be avoided, as instruments kept in such disinfectant solutions are not sterile, and therefore should not be used for surgical or other invasive procedures in which they may come in contact with sterile tissue. True cold sterilization requires use of a chemical sterilant product, meticulous cleaning of all instruments prior to immersion, and careful management of the solution itself to ensure that adequate concentration and contact time are achieved. If clean, non-sterile instruments are periodically required for minor procedures, these should not be kept in a disinfectant solution, but rather in a clean, dry, closed container. Once used, instruments should only be returned to the container after they have been thoroughly cleaned, undergone at least low-level disinfection (or higher depending on intended use as per Spaulding criteria, see Table 3), and completely dried.

Glass bead sterilizers, which are sometimes used in laboratory settings when performing multiple procedures on rodents in series, should not be used for quick sterilization of instruments in clinical practice, as these devices only sterilize the tip of the instrument and increase the risk of thermal tissue damage due to use of hot instruments.

Assessment of sterilization – Autoclaves

Quality control testing of autoclaves should be performed regularly and documented. For other methods of sterilization, consult the equipment manufacturer regarding specific means and frequency of quality control testing required.

- **Internal indicator strips** should be placed in every surgical pack prior to autoclaving. External autoclave indicator tape is not a reliable indicator of the sterility of a pack's internal contents. The external indicator tape should be evaluated by the individual removing packs from the autoclave, and the internal indicator strip should be evaluated by the individual opening the pack at the time of use.
- **Biological sterility indicators** should be used periodically. These indicators contain bacterial spores, which are the most resistant form of bacteria. After being autoclaved, the indicator is placed in a small bench-top incubator to ensure that all of the spores have been killed by the sterilization process. In human healthcare facilities it is recommended that these indicators are used daily. Weekly or bi-weekly use is likely adequate in most veterinary clinics, depending on how heavily the autoclave is used. A biological sterility indicator should also be used in the first cycle after the autoclave has been moved, repaired, or if there has been any indication of sterilization failure.

If internal or external indicators fail, the items in question must not be used and the event investigated immediately. In the event of biological indicator failure (with or without external or internal indicator strip failure), any implantable items (e.g. fixation plates, pins, screws) sterilized after the last biological indicator was tested and passed must be immediately recalled. Recall of other items is prudent but not considered necessary while the investigation is underway. Three more consecutive autoclave runs should be performed with biological indicators, and if any are positive, then all items processed since the last successful biological indicator run must be recalled and re-processed (Rutala 2017).

With any possible autoclave failure, the autoclave should be promptly serviced by certified personnel. Biological indicator results should be recorded in an autoclave log to allow for tracking of autoclave function. Autoclaves should be serviced regularly by certified personnel as part of a preventive maintenance schedule.

Labeling and storage

Label sterilized items with the date of sterilization, and ideally also the load number, to allow for removal of items of concern if problems are identified at a later date and for consideration of shelf-life. Do not write on the paper side of packaging if peel pouches are used, due to the risk of compromising package integrity. Store sterilized items in well ventilated areas that prevent accumulation of moisture, dust and extremes in temperature and humidity (Rutala & Weber 2009). Items should be minimally handled after sterilization to decrease the risk of unapparent damage to the packaging. Formal expiry times are not available for autoclaved items, however, one year is a reasonable limit for items that are stored and handled properly. Transportation of sterilized items between facilities is discouraged because of the potential for damage or loss of packaging integrity during transportation and handling.

References

Bailey KE, et al. Equine rotaviruses – Current understanding and continuing challenges. Vet Microbiol. 2013:167(1-2):135-144.

Block SS, ed. Disinfection, sterilization and preservation. 5th ed. Philadelphia: Lippincott, Williams & Wilkins. 2001.

Donkers LE, et al. Enterobacter cloacae epidemic on a neonatal intensive care unit due to the use of contaminated thermometers. Ned Tijdschr Geneeskd. 2001;145(13):643–647.

Hood E, et al. Flash sterilization and neurosurgical site infections: Guilt by association. Am J Infect Control 1997;25:156.

Hornig KJ, et al. Evaluation of the efficacy of disinfectant footmats for the reduction of bacterial contamination on footwear in a large animal veterinary hospital. J Vet Intern Med. 2016;30(6):1882-6. PubMed PMID: 27731908.

Jefferson JA. Central services. In: Association for Professionals in Infection Control, ed. APIC text of infection control and epidemiology. Washington, DC: APIC, 2009:55/51-18.

Linton AH, et al. Practical aspects of disinfection and infection control. In: Linton AH, Hugo WB, Russell AD, eds. Disinfection in veterinary and farm animal practice. London: Blackwell Scientific Publications. 1987;144-167.

Mangram AJ, et al. Guideline for prevention of surgical site infection, 1999. Hospital Infection Control Practices Advisory Committee. Infect Control Hosp Epidemiol. 1999;20:250-78.

Murphy CP, et al. The prevalence of bacterial contamination of surgical cold sterile solutions from community companion animal veterinary practices in southern Ontario. Can Vet J. 2010;51:634-636.

Public Health Ontario (PHO). Provincial Infectious Diseases Advisory Committee (PIDAC). Best practices for environmental cleaning for prevention and control of infections in all health care settings. 3rd ed. Toronto, ON: Queen's Printer for Ontario, 2018. Available at: https://www.publichealthontario.ca/-/media/documents/bp-environmental-cleaning.pdf?la=en. Accessed Dec-2018. Accessed Dec-2018.

Public Health Ontario (PHO). Chlorine dilution calculator. Available at: https://www.publichealthontario.ca/en/health-topics/environmental-occupational-health/water-quality/chlorine-dilution-calculator. Accessed Dec-2018.

Rutala W, Weber D. Infection control: The role of disinfection and sterilization. J Hosp Infect. 1999;43 Suppl:S43-55. PubMed PMID: 10658758.

Rutala WA, et al. Patient injury from flash-sterilized instruments. Infect Control Hosp Epidemiol. 1999;20:458.

Rutala WA, Weber DJ. Cleaning, disinfection and sterilization. In: Association for Professionals in Infection Control, ed. APIC text of infection control and epidemiology. Washington, DC: APIC. 2009;21/21-27.

Rutala WA, et al. Efficacy of improved hydrogen peroxide against important healthcare-associated pathogens. Infect Control Hosp Epidemiol. 2012;33(11): 1159-1161. PubMed PMID: 23041817.

Rutala WA, Weber DJ. Selection of the ideal disinfectant. Infect Control Hosp Epidemiol. 2014:35(7):855-65.

Rutala WA, Weber DJ. Disinfection, sterilization, and antisepsis: An overview. Am J Infect Control. 2016;44(5 Suppl):e1-e6. PubMed PMID 27131128.

Rutala WA, et al. Guideline for disinfection and sterilization in healthcare facilities, 2008 (update 15-Feb-2017). Centers for Disease Control and Prevention (CDC). Available at: https://www.cdc.gov/infectioncontrol/guidelines/disinfection/index.html. Accessed Dec-2018.

Serbetci K, et al. Effects of resterilization on mechanical properties of polypropylene meshes. Am J Surg 2007;194:375-379.

Spaulding E. The role of chemical disinfection in the prevention of nosocomial infections. In: Proceedings of the International Conference on Nosocomial Infections, 1970. Chicago, IL: American Hospital Association; 1971;247-54.

Stuetzer B, Hartmann K. Feline parvovirus infection and associated diseases. Vet J. 2014:201(2):150-155.

Weese JS. (2015). Cleaning and disinfection of patient care items, in relation to small animals. Vet Clin North Am Small Anim Pract. 2015;45(2):331-42. PubMed PMID PMID: 25542614.

Laundry and Waste Management

Laundry management

Although single-use, disposable linens and similar items are ideal from an infectious disease control aspect, such items can also produce tremendous waste. Laundry is therefore a very important component of infectious disease control in the clinic setting. Although soiled linens are a potential source of microorganisms, with appropriate hygienic handling, storage and processing of clean and soiled linens, the risk of disease transmission from these items can be reduced to an almost negligible level.

Linens and special clothing used in veterinary clinics (e.g. cage blankets, towels, surgical drapes, surgical gowns, scrubs, lab coats) can be an important means of moving pathogens from one area to another within the clinic, and to areas outside the clinic. As a result, **clinic clothing (e.g. scrubs, lab coats) should always be laundered on-site or sent to a commercial laundry facility that is equipped to handle laundry from medical/veterinary facilities.** This helps to prevent transmission of pathogens to family members, family pets and the general population. Personnel should change into clinic clothes at the beginning of their shift and back into street clothes at the end of their shift. Clinics should have appropriate laundry facilities or laundry services to accommodate the need to change clothing daily, or more frequently if required.

Microbial numbers on soiled linens (e.g. towels, blankets) and clothing are significantly reduced by dilution and during the mechanical action of washing and rinsing. Linens used in veterinary clinics should be laundered using detergent and dried in a hot air dryer to promote killing of microorganisms. Specific recommendations for air and water temperature for medical laundry are available (Sehulster 2015).

Linens contaminated with gross organic material must be pre-cleaned by hand to remove such material prior to laundering. Appropriate personal protective equipment (PPE) should be worn for this procedure (e.g. lab coat and gloves). It is not possible to adequately clean laundry by machine when gross organic material is present, and laundering such items can lead to contamination of other laundered items. In situations where laundry cannot be adequately cleaned of gross contamination, it should be discarded.

Collection and handling

Except for linens potentially contaminated with infectious agents (see below), all used linens can be handled in the same way. Heavily soiled linens should be rolled or folded to contain the heaviest contamination in the centre of the bundle, without contaminating personal clothing or the environment. Remove large amounts of solid debris, feces or blood clots from linens with a gloved hand and disposable tissue or paper towel, which are then immediately placed in the garbage or appropriate biomedical waste receptacle. Excrement should not be removed by spraying with water or shaking as this may result in contamination of the surrounding area and personal clothing. Contain wet laundry prior to removal from the immediate area or placement in the laundry bag by wrapping it in clean towels or sheets (PHO 2018), or placing in a plastic bag or bin, if necessary.

Linens contaminated with gross organic material must be pre-cleaned by hand to remove such material prior to laundering.

Key points:

- Handle linens with minimum agitation and shaking.
- Always place soiled linens directly in a hamper or bag designated for dirty laundry.
- Never place soiled linens on the floor.
- Tie laundry bags securely and do not over-fill.
- Always perform hand hygiene after handling soiled linens, including after glove removal.
- Clean carts and hampers after each use.
- Wash laundry bags after each use (they can be washed in the same cycle as the linens they contain).

Laundry should not be considered clean until it has been dried completely, ideally using the highest heat possible.

Transport & storage

Contaminated laundry in critical areas such as surgery should be kept in closed bins within a bag that can then be removed and brought to the laundry room at the end of the day or as required. Linens transported by cart should be moved in such a way that the risk of cross-contamination is minimized (e.g. avoid moving the cart from potentially contaminated areas (e.g. runs/ kennel area) to cleaner areas (e.g. prep room, surgery). If soiled laundry is significantly wet, a plastic garbage bag around the laundry bag should be used to contain any fluid that may penetrate the bag. This bag should be discarded when wet.

Clean linens should be transported and stored in a manner that prevents contamination. If laundry carts are used, separate carts should be used for clean and dirty linens. Clean laundry should kept in a designated area in cupboards with doors that can be closed to reduce risk of contamination. Ideally clean laundry should not be stored in the laundry room where it may become contaminated by incoming soiled laundry.

Washing and drying

Use of normal machine washing with a commercial laundry detergent and machine (hot air) drying are sufficient to greatly reduce the numbers of most significant infectious pathogens on most soiled linens (Sehulster 2015). The following points should be considered when washing and drying clinic laundry:

- If laundry is washed in cold water, an appropriate cold-water detergent must be used according to label directions.
- Do not assume that hot water washing will disinfect or sterilize items. Traditional high-temperature washing (71°C for 25 minutes or more) can significantly reduce bacterial numbers (Sehulster 2015), but standard household washing machines do not typically reach this temperature, even if the hot water setting is used.
- The heat and drying effects of tumble/hot air drying are a critical step in the laundering process, and account for a large proportion of the decrease in bacterial counts achieved. Therefore, laundry should not be considered clean until it has been dried completely, ideally using the highest heat possible.
    - Line-drying linens outdoors may have the advantage of also exposing the surface of the fabrics to ultraviolet (UV) light, if they are hung to dry in the sun. However, it is difficult to expose all surfaces to sunlight, and thick fabrics, items made of multiple fabric layers and those containing seams may protect bacteria from UV exposure. Also, the antimicrobial action of the high heat of tumble drying is lost if linens are line-dried, therefore tumble drying is recommended, especially for any materials that may have been contaminated with a transmissible infectious pathogen.

Laundry from infectious cases

Laundry from potentially infectious cases should be handled separately from other laundry using the following precautions:

- Collect contaminated linens in a separate laundry bag, and wash and dry them separately.
- For linens with gross contamination of a potentially infectious nature (e.g. feces from a diarrheic animal, discharge from an infected wound, urine from an animal with a urinary tract infection), remove as much organic material as possible by hand (using gloves and disposable tissue or paper towel, as described above). Then pre-soak the items in bleach solution (9 parts water:1 part household bleach) for 10-15 minutes prior to machine washing.
- Add bleach to the household detergent in the washing machine as per label instructions.
- Tumble dry all items on the highest heat setting available.

Safety of clinic personnel

Personnel need to protect themselves from potential transmission of pathogens from soiled linens by wearing appropriate personal protective equipment (e.g. gloves, gown, apron) when handling these items. Personnel should perform hand hygiene whenever gloves are changed or removed, or if they come in contact with soiled linens while not wearing gloves. Hand hygiene stations should be available in the laundry area.

Even though sharps disposal should occur at time of use, personnel need to be aware of the risk of sharps in the laundry, especially in the pockets of scrubs or lab coats. If a sharp is identified in the laundry, the incident should be reported to management and documented to prevent reoccurrence (PHO 2018).

Commercial laundry facilities

A company that specializes in handling laundry from medical/veterinary facilities should be used if it is not possible for laundry to be cleaned on-site. Adequate separation of clean and dirty laundry in the transport truck is essential to ensure that there is no opportunity for mixing or cross-contamination of clean and dirty linens.

Waste management

Veterinary biomedical waste is a potential source of both zoonotic and non-zoonotic infectious pathogens. Therefore, it is important to handle all such waste appropriately. In Canada, biomedical waste is defined and regulated at the provincial/territorial and municipal levels (usually by the applicable Department/Ministry of the Environment). Biomedical waste typically includes sharps, tissues (anatomic waste), highly contaminated (e.g. blood-soaked) materials, and dead animals. The Canadian guidelines for biomedical waste are available online (CCME 1992), but individual jurisdictions may have more stringent regulations. Details are typically available through provincial/state and municipal web sites, or through local veterinary regulatory bodies. It is important to ensure that all staff who may handle biomedical waste are aware of the relevant requirements in their area, and the information should be made readily accessible (e.g. documented in or linked to the clinic infection control manual). Small clinics in rural areas may be able to make arrangements with a local human hospital or other healthcare institution to have their waste disposed with that of the human facility, if biomedical waste disposal services are not otherwise available.

Although it is beyond the scope of these guidelines to describe veterinary biomedical waste management in detail, the following basic information may be helpful:

- **Used sharps** are considered biomedical waste and should be disposed accordingly. Use approved, puncture-resistant sharps disposal containers to remove, store and dispose of sharps such as needles, blades, razors and other items capable of causing punctures.
- **Non-anatomical waste saturated or dripping with blood** (e.g. blood-soaked lap sponges and gauze) are also best disposed of as biomedical waste.
- **Liquid waste** such as chest fluid, abdominal fluid, irrigating solutions, suctioned fluids, excretions and secretions usually may be poured carefully down a toilet or any drain connected to a sanitary sewer or septic tank. Jurisdictional regulations may dictate the maximum volume of blood or body fluids permitted to be poured into the sanitary sewer and whether pre-treatment (e.g. with bleach or disinfectant) may be required prior to disposal. If there is likely to be splashing during this disposal process, appropriate personal protective equipment should be worn.
- **All other waste,** such as general office waste and non-sharp medical equipment, may be disposed of in the regular waste stream, and requires no special treatment other than containment during disposal and removal.

Waste should be contained in a leak-proof container or bag that can be discarded with the waste (e.g. a plastic garbage bag of appropriate colour/transparency for the type of waste).

Urine and feces are not considered biomedical waste, nor is disposable equipment that has come in contact with an infectious animal (e.g. examination gloves, disposable gowns, bandage materials that are not saturated with blood). Nonetheless, some of these materials may pose a risk to clinic personnel, patients and waste disposal personnel in terms of their potential to transmit infectious pathogens. Therefore, additional precautions should be taken to minimize contamination of the clinic environment and the risks to people and animals from potentially infectious waste, whether it is considered biomedical waste or not. Precautions may include double-bagging of materials from isolation areas, keeping waste cans covered to prevent access by curious animals and to prevent spillage if a waste can is knocked over. If contamination of the inside of a waste can occurs (e.g. due to a tear in a garbage bag), the container should be thoroughly cleaned (and disinfected if needed) after emptying.

Waste bins are often required in examination rooms. If bins are in the open and do not have a fitted lid or contain potentially infectious or messy waste, they should be emptied between patients so they are not investigated or knocked over by subsequent patients. Otherwise waste bins should generally be emptied at the end of the day.

References

Canadian Council of Ministers of the Environment (CCME). Guidelines for the management of biomedical waste in Canada. 1992. Available at: http://publications.gc.ca/collections/collection_2015/ec/En108-3-1-42-eng.pdf. Accessed Dec-2018.

Public Health Ontario (PHO). Provincial Infectious Diseases Advisory Committee (PIDAC). Best practices for environmental cleaning for prevention and control of infections in all health care settings. 3rd ed. Toronto, ON: Queen's Printer for Ontario, 2018. Available at: https://www.publichealthontario.ca/en/eRepository/Best_Practices_Environmental_Cleaning.pdf. Accessed Dec-2018.

Sehulster LM. Healthcare laundry and textiles in the United States: Review and commentary on contemporary infection prevention issues. Infect Control Hosp Epidemiol. 2015;36(9):1073-88. PubMed PMID: 26082994.

Special Procedures

Surgery

All surgical procedures cause breaks in the normal defensive barriers of the skin or mucous membranes. These breaks are accompanied by an inherent risk of surgical site infection (SSI). Surgical site infections can occur sporadically or as part of an outbreak, and can have devastating outcomes in some situations. A variety of pre-, intra- and post-operative factors can influence SSI risk, and prevention of SSIs involves use of a range of measures. Good general infection control practices (e.g. hand hygiene, cleaning and disinfection) are important for prevention of SSIs. Specific measures pertaining to surgery include maintenance of the surgical environment, use of appropriate personal protective equipment and hand hygiene, disinfection and sterilization of anesthetic equipment and surgical instruments, appropriate use of perioperative antimicrobials, and surgical site care before, during and after the procedure. Many of the recommendations below are already considered minimum practice standards in various jurisdictions, but actual requirements may vary. Veterinarians should contact their veterinary regulatory authority for details about the specific regulations in their area.

Risk factors for the development of SSIs in veterinary surgery include surgery classification (clean vs. dirty), duration of surgery and aesthesia, patient characteristics, and other pre-, intra-, and post-operative factors (Verwilghen & Singh 2015). Not all of these factors are modifiable but it is nonetheless important to take all reasonable precautions to reduce the risk of SSI development in veterinary patients.

Surgical environment and suite design

Having a well-designed and maintained surgical area or suite is very important. In order to keep the surgical environment as clean as possible, this area should be separated from personnel and animal traffic, and all surfaces should be easy to thoroughly clean and disinfect. **A surgical suite should only be used for one surgical procedure at a time** and should not be used for non-surgical procedures between surgeries. The surgical suite should be designed so that the procedure can be efficiently performed and personnel can effectively work and move around in the room without compromising the sterile field. Entrance to the area should be restricted at all times to minimize traffic in the room. The number of people in the surgical area has been identified as a risk factor for SSI in small animals, so only essential personnel should be allowed in the area during any surgical procedure. All personnel participating in the procedure, including those performing surgical nursing duties, must be trained in operating room procedures.

Surgical suites should be dedicated rooms that are fully enclosed with walls running to the ceilings and no outer windows. Sinks should not be present in the surgical suite. In contrast to human hospitals, some veterinary hospitals have multiple surgical tables in operating rooms. This arrangement raises concern about cross contamination, as well as potential risks posed by the greater number of people in the room and more frequent movement in and out of the room. Performing more than one surgical procedure at a time in the same operating room should be discouraged, especially procedures that might be associated with aerosolization of bacteria (e.g. dental procedures).

Air handling

Air movement within an operating room should be organized, predictable and consistent, to maintain the principle of clean-to-dirty air movement. Standards recommended for human hospitals are not commonly achieved in veterinary hospitals but they should be considered a standard to strive for in any newly designed facility. These include:

- positive pressure ventilation (positive pressure in the operating room with respect to adjacent rooms and corridors).
- minimum of 15 air exchanges per hour (at least 3 of which should be fresh air).
- use of appropriate air filters.
- introduction of air at ceiling level with exhaust near the floor.
- operating room doors must never be propped open.
- ventilation of operating room at all times.

Basic aspects such as the level of air introduction and exhaust should be provided for all operating rooms. Window air conditioners, windows with drafts, incompletely enclosed rooms and similar issues that disrupt organized airflow are inadvisable.

The clinical efficacy of ultraclean air rooms, air filtration or treatment, laminar airflow technology, ultraviolet light treatment of air, and other methods such as disinfectant misting or fogging are not currently recommended for routine use as there is insufficient clinical evidence of their effectiveness.

Stocking the surgical suite

Commonly used surgical supplies should be stored within the operating room or in an adjacent room in a manner that prevents contamination by direct contact or aerosols (e.g. in a closed cabinet). If they are not stored in the operating room, items required for a particular procedure should be transported to the operating room immediately before the procedure. These items should be transported on a covered cart or in a case with the cover that can be removed once the items are delivered. Non-surgical items should not be stored in the operating room in order to minimize traffic in the room.

Surgeon preparation areas

Surgical personnel must be able to perform hand antisepsis in close proximity to, but not within, the operating room, to prevent droplet and aerosol contamination of the environment generated during scrubbing with soap and water or other preparation activities. The use of an alcohol-based surgical hand rub (ASHR) significantly reduces this contamination risk (see preoperative hand antisepsis section below for more information on ASHRs). ASHRs are also highly beneficial in clinics without the necessary infrastructure (e.g. physical layout and sinks) to perform soap and water scrub effectively in proximity to the surgical suite. If veterinarians prefer a soap and water hand scrub, sinks should be located away from areas of heavy traffic, areas where patients are housed or prepared for surgery, and open shelving where sterile supplies are stored. Scrub sinks should be designed so that all required supplies are readily accessible and the operating room can be entered without risk of hand contamination.

Surgical waste

Be sure to follow all local regulations pertaining to biomedical waste (see Chapter: Laundry and Waste Management). While the risk of zoonotic infection from blood or tissues from companion animals is relatively low, nonetheless all blood or tissue contaminated items must be managed as potentially biohazardous.

Personnel considerations

Preoperative hand antisepsis

Alcohol-based surgical hand rubs (ASHRs): ASHRs are now the recommended method for surgical hand antisepsis in human medicine (Pittet 2009), but are less common in veterinary clinics. These products have a rapid-kill effect due to their high alcohol content. ASHRs take less time to apply, are less irritating to the skin (particularly with repeated use), and have been shown to be as effective as soap-and-water scrubs for reducing bacterial flora on the hands, when correctly applied (Widmer 2010). It is not necessary for ASHR to contain additional active ingredients, such as chlorhexidine gluconate (CHG) or ortho-phenylphenol, as these products do not enhance the efficacy or residual activity of the product (Kampf 2017). Hand washing with a neutral soap is recommended prior to the first use of an ASHR on a given day, and anytime hands are visibly or likely soiled (e.g. after handling a patient between procedures). Hands must be completely dry before application of the ASHR. Always follow the manufacturer's directions regarding the amount of ASHR product to use and application technique. General guidelines for use of an ASHR are as follows (WHO 2009):

- Remove all hand and arm jewelry, including rings, bracelets and watches.
- Use a pick or file to clean all dirt out from underneath the fingernails.
- If hands or arms are visibly dirty, initially wash with soap and water as per regular hand hygiene protocols.
  - Ensure hands and arms are dry before applying ASHR.
- Use approximately 15ml of product to ensure that hands remain wet throughout the procedure.

- Dispense product into one palm using the elbow of the opposite arm, or a foot pump if available.
- Rub fingertips into solution for at least 5 seconds to clean under the nails.
- Rub product onto the forearm up to the elbow until skin is dry.
- Repeat for opposite arm.
- Dispense additional product into one hand and then rub both hands together until dry, ensuring all surfaces of the hand are covered, including palms, backs of hands, between fingers, back of fingers, and base of thumbs.
- Entire procedure should take approximately 1.5-2 minutes.

Soap-and-water surgical hand scrub: A surgical hand scrub should be performed before donning a sterile gown and sterile gloves. Various surgical scrub techniques have been described (WHO 2016). Most commonly, a structured five-minute surgical scrub with antibacterial soap is used. There is no indication that there is any benefit to a scrub lasting longer than five minutes, and scrubbing for longer than necessary can cause damage to the skin and increase discomfort and the risk of skin infection. Always use warm water, as hot water is irritating to the skin (WHO 2009).

An example of a standard soap-and-water surgical scrub is as follows; further details can be found in currently available veterinary surgery textbooks (Fossum 2013 Johnston & Tobias 2018):

- Remove all hand and arm jewelry, including rings, bracelets and watches.
- Use a pick or file to clean all dirt out from underneath the fingernails.
- If hands or arms are visibly dirty, initially wash with soap and water as per regular hand hygiene protocols.
- Lather both hands and forearms with antibacterial soap.
- Using a soft bristled brush, scrub fingers, palms, back of hands, wrists and forearms to just below the elbows from distal to proximal (i.e. cleanest to dirtiest), alternating from left to right for each part. Pay special attention to fingertips, nail beds, and finger/thumb webs.
- Thoroughly rinse off all soap with clean running water, while keeping the hands above the elbows to ensure all water runs off at the elbows and not the fingers.
- Use a sterile towel to dry the hands and arms completely, proceeding from distal to proximal (i.e. fingers to elbows), before donning a gown and gloves.

The most commonly used soaps for surgical hand scrubs contain either CHG or povidone-iodine (PI). CHG soap has been shown to be more effective for reducing bacterial counts on the hands than PI soap in some studies (Tanner 2008), although there are more concerns about skin irritation from use of CHG versus PI.

Scrubs worn in surgery should not be worn when handling or treating other patients, and at a minimum should be covered with a lab coat when outside the surgery area.

Personal protective equipment

All personnel in the surgical area should wear designated surgical scrubs, a surgery cap or hair bonnet, and a nose-and-mouth mask when surgery is underway, regardless of whether or not they are directly involved in the procedure itself. Scrubs worn in surgery should not be worn when handling or treating other patients, and at a minimum should be covered with a lab coat when outside the surgery area (see Chapter: Personal Protective Equipment). Personnel directly involved in the procedure should also wear a sterile gown and sterile gloves. It is important to follow appropriate surgical gowning and gloving techniques that prevent contamination of any part of the gown or gloves that may come in direct contact with patient's tissues or any part of the sterile surgical field or instruments. Common methods are briefly described here; further details can be found in currently available veterinary surgery textbooks (Fossum 2013, Johnston & Tobias 2018).

Gowning technique:

- Have the folded gown set out on a sterile surface within the surgical suite
- Hands should only come in contact with interior surfaces of the gown; be familiar with how the gown is folded in order to avoid accidental contamination of the outer surfaces
- Grasp the gown and lift it away from the table to provide adequate room for gowning
- Allow the gown to unfold without shaking it, in order to decreases the risk of contamination of the gown or disturbing dust particles in the room
- Slide arms through arm holes, keeping hands inside the cuffs of the gown
- Have the neck ties secured by an assistant. Only secure front ties (if necessary) after donning sterile gloves, with or without assistance depending on the style of gown.

Glove donning techniques:

- Closed gloving (can only be performed after donning a sterile surgical gown)(for a demonstration video see PennVet 2013)
  - With hands still within the cuff of the surgical gown, pick up one glove and place it with thumbs and fingers pointing toward the elbow, palm side down, on the corresponding palm
  - Grasp the cuff on the underside of the glove with the thumb and index finger of the hand to be gloved (through the cuff of the gown), while using the free hand to grasp the cuff on the other side of the glove
  - Use the free hand to pull the cuff of the glove over the opposite hand and the cuff of the gown, while pushing the hand up into the glove. Ensure that the glove covers the entire cuff of the gown when complete.
  - Repeat with other hand. Ensure that the already gloved hand does not come in direct contact with the cuff of the gown of the opposite hand.
- Open gloving (can be used for non-surgical procedures that require sterile gloves but not a sterile gown, or for replacing a contaminated glove during surgery) (for illustrated instructions see Alberta Health Services 2016)
  - Pick up one glove by the inside surface of the folded cuff and pull over the opposite hand until fingers are in the fingers of the glove
  - Hook the folded cuff of the glove over the thumb by anchoring thumb inside near the thumb of the glove
  - Repeat with other hand.
  - Place the gloved fingers of one hand under the edge of the cuff of the glove and pull it down around the wrist, while pushing the fingers the rest of the way into the glove. If necessary, then grasp the thumb of the glove from the outside to allow the thumb on the inside to be repositioned in the thumb opening. The outside surface of one glove should only ever touch the outside surface of the opposite glove.
  - Repeat with other hand.
  - Ensure that the entire cuff of the surgical gown if covered by the cuff of the glove, if applicable
- Assisted Gloving
  - Have a sterile assistant pick up the glove under the cuffs
  - As they hold it open with the thumb of the glove facing the surgeon, push hand into glove
  - Have the assistant gently release the glove cuff such that the cuff of the gown is completely covered
  - Repeat with other hand

Foot covers (booties) are not required if footwear is clean. However, dedicated clean surgical footwear is ideal to minimize contamination of the surgical environment. All footwear must be closed-toed.

Glove perforation: Glove punctures during surgery are common, especially during procedures lasting an hour or more, and often go unnoticed (Verwilghen & Singh 2015). Although the resulting risk of SSI in veterinary patients is unclear, glove perforation has been associated with increased risk of SSI in human patients (Verwilghen & Singh 2015). **Glove use, therefore, does not negate the need for proper preoperative hand antisepsis.** Double gloving can be used to provide an additional layer of protection.

While double gloving does not decrease the overall incidence of glove perforation, punctures of both the inner and outer layers are less common. Double gloving can also be used to reduce contamination of the surgical field due to accidental

contamination of gloves during patient draping and the initial incision, by removing the outer gloves after these steps. This method is often advocated for procedures that have a higher risk of SSI, such as orthopedic surgery involving implants (Verwilghen & Singh 2015).

If glove perforation is identified, gloves should be changed immediately unless the short delay required would result in surgical complications. When double gloves are used, the inner glove must be carefully inspected following removal of the outer glove to ensure that there was not perforation of both gloves. The outer glove can then be replaced if warranted.

Equipment considerations

Sterilization of instruments

Complete sterilization of surgical instruments and any other items that might come in contact with the surgical field is critical to preventing SSIs. Poor sterilization or inappropriate handling of instruments after sterilization can result in contamination of sterile tissues during surgery. Outer indicator tape and inner indicator strips should be checked as soon as a surgical pack is opened, prior to use. If there is any indication of failed sterilization, use a new pack and investigate autoclave function. Biological indicators are also important quality control measures. Inner indicator strips and biological indicators should be routinely used to keep record of autoclave function.

Immediate-use steam sterilization (i.e. "flash" sterilization) is a rapid sterilization method performed on unwrapped items. It is designed for rare situations where there is an immediate need for sterilization, such as when a critical instrument is dropped during surgery and there is no replacement. This method should **never** be used for surgical implants. It should also not be used in lieu of proper surgical planning or purchase of adequate numbers of instruments (PHO 2013).

(see Chapter: Cleaning, Disinfection, Sterilization for more information regarding sterilization techniques)

Disinfection of anesthetic equipment

Endotracheal tubes: In human medicine, endotracheal (ET) tubes are typically considered single-use devices, but reuse of ET tubes has become more common with the rising costs of healthcare. These tubes are considered semi-critical equipment, and as such should be subjected to high-level disinfection or sterilization between patients. They can be effectively sterilized using glutaraldehyde or ethylene oxide gas, although the physical integrity of the cuffs in particular can be compromised by repeated sterilization with these methods. In veterinary medicine, it is impractical to discard ET tubes after a single use, but chemical sterilization may not be readily available. Evidence-based guidelines for reuse of ET tubes in veterinary medicine are not available. Nonetheless, **at an absolute minimum,** ET tubes must be thoroughly cleaned (inside and outside) with hot water and detergent immediately after use to prevent any discharge or debris from drying and forming a biofilm on the device. Tubes should then be soaked in a solution of accelerated hydrogen peroxide (AHP) or CHG for at least 5 minutes, rinsed thoroughly and dried prior to being reused (Crawford & Weese 2015). It is important to test the integrity of the cuff before every use to ensure the device has not been compromised by repeated cleaning and disinfection.

Anesthetic gas tubing and rebreathing bags: Although the tubing connecting the anesthetic machine to the patient's endotracheal tube should not come in direct contact with the patient, moisture and condensation often accumulate in the tubes and may contain microorganisms from the animal's airway. In human medicine, this equipment is also typically single-use. As for ET tubes, evidence-based guidelines for reuse of this equipment in veterinary medicine are not available. **At a minimum,** gas tubing should routinely be thoroughly washed with hot water and detergent and hung to dry at the end of the day's procedures, or more often if they are heavily used. If there is visible discharge in the tubing, or if the animal has a known or suspected respiratory tract infection, the tubing should be thoroughly cleaned with hot water and detergent, soaked in a solution of AHP or CHG, rinsed with water and dried prior to being reused. The corrugations should be routinely checked for accumulation of debris or mold. Rebreathing bags should be cleaned and disinfected as for the associated gas tubing, as they also come in contact with the expired air from the patient.

If an animal has a known or suspected transmissible respiratory tract infection, filters are available which can be placed between the ET tube and the rest of the anesthetic circuit in order to help protect the equipment from contamination.

Perioperative antimicrobials

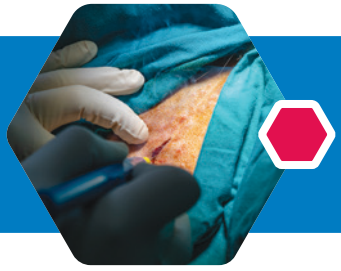
Administration of perioperative (i.e. before, during and after surgery) antimicrobials is an important and complex issue. The goal of perioperative antimicrobial therapy is to reduce the risk of postoperative infection, while minimizing the negative impact on the patient's natural microbiota and the risk of antimicrobial-associated complications such as diarrhea, and the emergence of antimicrobial resistant bacteria. Surgical site management and infection prevention protocols are important tools to help reduce the need for perioperative antimicrobial use (see Chapter: Antimicrobial Stewardship for more information and resources).

There is currently little objective information about the need for antimicrobials for specific veterinary procedures, as well as the optimal choice of drug(s), timing and dosages. **Antimicrobials are indicated in many clean-contaminated, contaminated and dirty procedures.** Descriptions and examples of surgical wound classifications can be found in Table 1 below. Antimicrobial prophylaxis for clean procedures is generally considered unnecessary, but is still relatively common in veterinary medicine. In human medicine, antimicrobials are not typically recommended for clean procedures (e.g. arthroscopy). Regardless, it is unclear whether recommendations from human medicine should be directly extrapolated to veterinary procedures, because there are obvious differences in post-operative incision care and patient environment for animals, which may increase the risk of infection. It has become common practice in veterinary medicine to use antimicrobials for elective orthopedic surgeries such as tibial plateau leveling osteotomy (TPLO) or hip replacement, despite their designation as clean procedures. This is due to the high rates of SSIs associated with these procedures, the potentially catastrophic consequences of such infections, along with data indicating a protective effect of antimicrobials for TPLO (Verwilghen & Singh 2015). Concerns with this practice include inappropriate timing of administration (e.g. too far in advance of surgery or starting after surgery), excessive duration of therapy, inadequate dosing and inappropriate drug choice.

TABLE 1. Surgical wound classifications (adapted from Fossum 2013, Johnston & Tobias 2018, WHO 2018)

Classification	Description	Examples
Clean	Non-traumatic, non-inflamed operative wounds in which the respiratory, gastrointestinal, genitourinary, and oropharyngeal tracts are not entered	Exploratory laparotomy Elective neuter Total hip replacement
Clean-contaminated	Operative wounds in which the respiratory, gastrointestinal, or genitourinary tract is entered under controlled conditions without contamination; an otherwise clean wound in which a drain is placed	Cholecystectomy Small intestinal resection Soft palate resection
Contaminated	Open, fresh, accidental wounds; procedures in which gastrointestinal contents or urine is spilled or a major break in aseptic technique occurs	Open cardiac massage Cystotomy with spillage of infected urine Lacerations
Dirty	Old traumatic wounds with purulent discharge, devitalized tissue or foreign bodies; procedures in which a viscus is perforated or fecal contamination occurs	Excision or drainage of an abscess Peritonitis Perforated intestinal tract

When antimicrobials are used, therapeutic drug levels should be present throughout the *period of risk*. This starts at the time of initial incision and continues until the end the procedure, or a short time thereafter.



If perioperative antimicrobials are used every effort should be made to ensure that such use is as effective and evidence-based as possible. A key concept of peri-operative antimicrobial prophylaxis is that, when antimicrobials are used, therapeutic drug levels should be present throughout the *period of risk.* This starts at the time of initial incision and continues until the end the procedure, or a short time thereafter, since a fully functional barrier is not necessarily immediately achieved with incision closure. This typically requires parenteral administration of an antimicrobial within one hour of the first incision. If the surgical time is longer than two half-lives of the drug(s), then intraoperative redosing is indicated. In human medicine, it has been shown that starting antimicrobial therapy after surgery is no more effective than not using antimicrobials at all (Bratzler & Houck 2005).

The antimicrobial(s) selected for perioperative use should be appropriate for the patient, procedure, and expected opportunistic pathogens. Table 2 lists some of the most commonly used perioperative antimicrobials. It should be noted that for the vast majority of procedures cefazolin, cefoxitin, clindamycin, and ampicillin are likely appropriate. When administering perioperative antimicrobials parenteral routes are preferred, and oral administration is not recommended.

TABLE 2. Commonly used perioperative antimicrobials for small animal surgical procedures

Drug Name	Dosage	Half Life	Comments
Most Commonly Used			
Cefazolin	22mg/kg; q8-12hr IV, IM, SC	1hr	1st generation cephalosporin Gram-positive bacteria (e.g. *Staphylococcus, Streptococcus)* Some gram-negative (e.g. *Pasteurella, E. coli),* although resistance is common
Cefoxitin	Dogs: 30mg/kg; q12hr; IV Cats: 22-33mg/kg; q8hr; IV, IM	45-60min	2nd Generation Cephalosporin Anaerobes and gram-negative bacilli
Clindamycin	Dogs: 11mg/kg; q8-12hr; SC, IV Cats: 5-33mg/kg; q12-24hr; oral, SC	4-4.5hr (5.5mg/kg) 7-10hr (11mg/kg)	Gram-positive and anaerobic bacteria Often used for dental procedures in patients with periodontal disease
Ampicillin	10-20mg/kg; q6-8hr; IV, IM or SC	1-1.5hr	Gram-positive cocci and bacilli (e.g. streptococci, non-beta lactamase producing staphylococci) Many gram-negative bacteria are resistant
Less Common			
Metronidazole	10-12mg/kg; q8hr; IV	4-5hr	Anaerobic bacteria Dilute and give slowly over 20min if using IV
Gentamicin	Dogs: 4.4-6.6mg/kg; q24hr; IV, SC, or IM Cats: 5-8mg/kg; q24hr; IV, SC or IM	1-2hr	Gram-negative bacteria Doses for dogs up to 9-14mg/kg may be given to septicemia patients, if renal function is closely monitored
Amikacin	Dogs: 15-30mg/kg; q24hr; IV, IM or SC Cats: 10-14mg/kg; q24hr; IV, IM, or SC	1-2hr	Gram-negative bacteria, often used for serious infections Activity against some resistant bacterial strains

Typically, antimicrobials are not needed after surgery because the highest-risk time for contamination of the surgical site (i.e. during the surgery itself) is already passed. Recommendations in human medicine typically suggest cessation of antimicrobials within 24 hours postoperatively (Bratzler 2013). There is some evidence for use of antimicrobials post operatively for TPLO patients however, the optimal duration is not known. It is likely that there is little additional benefit after 48 hours of post-operative antimicrobials. More research is required to identify the ideal length of time for antimicrobial therapy after this and other similar procedures (Verwilghen & Singh 2015).

Surgical site management

Preoperative care

Preoperative management of the surgical site is very important, but there has been very little research in this area in veterinary medicine. The goal of preoperative surgical site management is to eliminate potential pathogens without creating a physical environment that may increase bacterial colonization or infection post-operatively. Maintaining a healthy skin barrier is critical. Preparation methods that damage the skin, such as excessive clipping, shaving or excessive scrubbing, must be avoided. Techniques for preoperative surgical site preparation and hand antisepsis tend to vary between clinics, and inappropriate practices, including short contact times and choice of preparation agents, are common (Anderson 2013). As such, veterinary personnel involved with patient preparation should be properly trained and educated to avoid unnecessary surgical site contamination.

Management of the patient's systemic condition (e.g. normothermia, glucose control, oxygenation, total anesthesia time) also play a role in prevention of SSIs, although these factors have not yet been closely examined in veterinary medicine.

If the patient's hair coat is visibly dirty, bathing the animal before surgery is reasonable as long as there is adequate time for the hair coat to dry before the procedure. In humans, it has been suggested that any method of hair removal can be associated with higher SSI rates, but obviously this cannot be avoided for the vast majority of procedures in veterinary medicine. Shaving the surgical site the night before has been associated with higher SSI rates in humans, therefore clipping (not shaving) of the surgical site should only be performed right before surgery. Care must be taken to avoid damaging the skin during this procedure, as abrasions provide sites for invasion and proliferation of opportunistic bacteria. The goal of clipping is not to remove all remnants of hair from the site, but rather to facilitate subsequent skin antisepsis by reducing the amount of hair. Excessive clipping can result in skin trauma, which is a risk factor for infection. Use of good quality, well-maintained clippers and blades helps to reduce the risk of skin abrasions (see Chapter: Cleaning, Disinfection, Sterilization for specifics on maintenance of clippers). If skin lesions around the surgical site are noted before or after surgery, the finding should be recorded and investigated, to determine whether equipment maintenance and/or personnel training need to be improved. **Do not clip animals in the surgery area/suite itself.** Use a "prep" area outside of the surgery area for this and any other preoperative procedures, including the first step of the skin preparation.

Skin preparation after clipping is also important. Typical practices include thorough cleaning and scrubbing of the site with antibacterial soap, followed by application of alcohol, and finally application of an antiseptic that is usually left on the skin. The final antiseptic application step should be performed after the animal has been positioned on the surgery table. As with all disinfectant and antiseptic products, manufacturer's recommendations regarding proper use, including contact time, must be followed in order to maximize efficacy. The two most effective and commonly used antiseptic agents for surgical site preparation are PI or CHG in either alcohol or aqueous solutions. However, CHG is irritating to mucous membranes and should be avoided when there may be exposed to mucous membranes (e.g. preparation of the head, near the eyes, nose or mouth). The application pattern should work from the cleanest area (i.e. incision site) outward to the dirtier areas (i.e. periphery of preparation area). The skin must be allowed to dry before the surgical procedure begins, particularly with use of products containing alcohol as these are flammable. Additional information on preoperative patient skin preparation is available in standard surgical reference texts (Fossum 2013, Johnston & Tobias 2018).

Preparation methods that damage the skin, such as excessive clipping, shaving or excessive scrubbing, must be avoided. Clipping (not shaving) of the surgical site should only be performed right before surgery.

In human medicine, there is evidence that the application of povidone-iodine solutions as a one-step paint or spray may be as effective as the traditional scrub-and-paint with the same solution (Elenhorn 2005), with the added benefit of shorter preparation time and reduced skin trauma. Extrapolation of this finding to veterinary patients must be done with caution, as scrubbing may be more necessary for animals if the haircoat and underlying skin are more heavily contaminated with dirt and debris than bare human skin.

Potential issues that need to be considered when developing clinic standard operating procedures for preoperative skin preparation include:

- preparing a large enough area of skin, in case extension of the intended incision is required.
- adequate initial cleaning with soap and water.
- potential for contamination of preparation solutions.
- ensuring adequate contact time with antiseptics.
- avoiding contamination of the surgical site during and after preparation.

Bandage changes should be performed using aseptic technique.

If skin preparation solutions (e.g. antibacterial soap and water, alcohol, chlorhexidine, iodine) are kept in refillable containers, these containers must themselves be cleaned and then disinfected when empty before being refilled. Contamination of these solutions with bacteria that are resistant to their respective antimicrobial actions can occur. Refilling the containers without disinfecting them can allow these resistant contaminants to accumulate. An outbreak of catheter site infections was reported in a small animal clinic that was associated with contaminated skin preparation solutions (Mathews 1996).

Surgical safety checklists

An emerging trend in human medicine is the use of **surgical safety checklists,** which have been demonstrated to reduce adverse events and errors, ultimately lowering surgery-associated morbidity and mortality. They help engage the entire surgical team in good communication and are an excellent tool to aid in the prevention of SSIs and other complications. See References for the World Health Organization (WHO) Surgical Safety Checklist. See the Appendix for a template veterinary surgical safety checklist that can be tailored to specific surgical environments and procedures.

Postoperative care

Postoperatively, a surgical incision site is highly susceptible to opportunistic infection from the bacteria of the patient's own microflora, from the environment or from hospital personnel. Avoid contact with the surgical incision, particularly with bare hands, as much as possible. Covering or bandaging incisions for 24 to 48 hours after surgery has been recommended in humans (Nicks 2010); this is also a reasonable recommendation in small animals in most situations, but can be considerably more challenging depending on the location of the incision, body shape and demeanor of the patient. Bandage changes should be performed using aseptic technique. Pet owners and handlers should be instructed on how to manage an animal with an incision, and the signs which may indicate the development of a SSI. There is no objective information about the need to cover surgical incisions for more than 48 hours in veterinary or human medicine, but preventing the animal from licking, scratching or otherwise traumatizing the surgical site is critical. Damage to the healing incision or the skin around it can result in the deposition of opportunistic pathogens, and make it easier for bacteria to grow in the area.

SSI surveillance

Every facility should have some form of a SSI surveillance program, as such programs have been shown to help reduce rates of hospital acquired infections. Even with relatively limited effort, sufficient data can be collected to provide a general understanding of endemic SSIs rates and any changes that may occur, allowing the veterinary team to review protocols and procedures related to SSI prevention (also see Chapter: Surveillance).

Accurate SSI rate surveillance is dependent on a number of factors, including accurate identification of SSIs (and differentiation from other infections), use of standard SSI definitions (see Table 3 below), the ability to determine procedure-specific surgical caseload numbers as denominators, and the ability to collect the data centrally and analyze them on a regular basis (ideally in real time). The most common active and passive SSI surveillance methods used (see Table 4) each have limitations that need to be considered. There is no standard approach for veterinary facilities, and factors such as caseload (number and types of procedures), number of clinicians and personnel availability must be considered when determining the optimal program. In general, active methods will provide the most accurate data but are also the most time consuming. No method is ideal, and facilities are encouraged to develop novel approaches to suit their needs (Burgess 2015). Methods such as automatic email questionnaires querying the health of the patient and describing SSIs sent to the owner at a pre-determined post-operative day (e.g. day 30) might be useful, albeit still with some limitations (e.g. response rate, reliance on owners to identify and characterize abnormalities).

Active surveillance programs tend to be most effective as SSIs are often diagnosed post-discharge, though they are more challenging to implement. Through actively identifying cases those patients with SSIs that may be superficial and not serious enough to warrant a recheck appointment are captured and a more accurate picture of SSIs can be generated. This can then be used to identify the areas of the infection control program that are effective or need improvements

TABLE 3. Definitions of different categories of surgical site infections (SSIs) (adapted from Johnston & Tobias 2018, NHSN 2019)

Category	Definition
Superficial SSI	Occurs within 30 days postoperative Affects skin and/or subcutaneous tissues of the incision Includes at least one: - Purulent discharge - Aseptically cultured bacteria - Diagnosis by the surgeon - Reopening of the incision accompanied by pain, heat, redness, swelling
Deep SSI	Occurs within 30-90* days postoperatively Affects deep soft tissues of the incision Includes at least one: - Purulent discharge - Reopening of incision spontaneously, or by surgeon if patient has pain, heat, redness, or tenderness - Abscess or other evidence of infection
Organ/Space SSI	Occurs within 30-90* days postoperatively Affects any area other than the incision that was encountered during the surgery Includes at least one: - Purulent discharge - Bacteria - Abscess or other evidence of infection

*depending on procedure, including those involving implants

TABLE 4. Examples of surgical site infection (SSI) surveillance methods

Method	Description	Comments
Medical record review	Periodic review of SSI reports in medical records	• Dependent on medical record quality. • Can underestimate SSI rates if patients with SSIs are seen by other clinics (e.g. referring veterinary, emergency clinic) and information is not conveyed to the surgical facility. • May be difficult to apply standard definitions to determine SSI with accuracy. • Retrospective, so early changes may be missed. • May require significant time and effort periodically depending on type of records and ability to search (e.g. paper vs electronic records).
Centralized collection of SSI reports	Real time reporting of SSIs detected during routine rechecks or communications to central clinic person	• Dependent on quality of reported data and accurate reporting by clinicians. • Can underestimate SSI rates if patients with SSIs are seen by other clinics (e.g. referring veterinary, emergency clinic) and information is not conveyed to the surgical facility. • Provides timely identification of infections and potential changes in SSI rates. • Fosters infection control communication. • Requires limited time and effort on a daily basis.
Active owner follow-up	Contacting owners at predetermined times specifically to identify SSIs (outside of routine follow-up calls)	• Most accurate data but labour intensive. • Can be made more practical using tools such as email surveys sent at a predetermined time post-operatively.

References

Alberta Health Services. Donning and doffing sterile gloves when a sterile gown is not worn. 2016. Available at: https://www.albertahealthservices.ca/assets/healthinfo/ipc/if-hp-ipc-glove-use-selection-donning-doffing-sterile-gloves-powerpoint.pdf. Accessed Dec-2018.

Anderson MEC, Foster BA, Weese JS. Observational study of patient and surgeon preoperative preparation in ten companion animal clinics in Ontario, Canada. BMC Vet Res. 2013;9:194.

Bratzler DW, Houck PM. (2005). Antimicrobial prophylaxis for surgery: an advisory statement from the National Surgical Infection Prevention Project. Am J Surg 2005;189:395-404.

Bratzler DW, et al. Clinical practice guidelines for antimicrobial prophylaxis in surgery. Surg Infect (Larchmt). 2013;14(1):73-156. PubMed PMID: 23461695

Burgess BA, et al. Veterinary hospital surveillance systems. Vet Clin North Am Small Anim Pract. 2015;45(2):235-42. PubMed PMID 25534534

Character BJ, et al. Postoperative integrity of veterinary surgical gloves. J Am Animal Hosp Assoc. 2003;39(3):311-20

Crawford S, Weese JS. Efficacy of endotracheal tube disinfection strategies for elimination of Streptococcus zooepidemicus and Bordetella bronchiseptica. J Am Vet Med Assoc. 2015;247(9):1033-6.

Ellenhorn JD, et al. Paint-only is equivalent to scrub-and-paint in preoperative preparation of abdominal surgery sites. J Am Coll Surg. 2005;201(5):737-41.

Fossum T, ed. Small Animal Surgery, 4th ed. St. Louis: Elsevier Mosby, 2013.

Girard R, et al. Tolerance and acceptability of 14 surgical and hygienic alcohol-based hand rubs. J Hosp Infect. 2006;63(3):281–8.

Hayes GM et al. Investigation of incidence and risk factors for surgical glove perforation in small animal surgery. Vet Surg. 2014;43(4):400-4.

Harris CL, et al. Best practice recommendations for the prevention and management of surgical wound complications. Canadian Association of Wound Care; 2018. Available at: https://www.woundscanada.ca/docman/public/health-care-professional/bpr-workshop/555-bpr-prevention-and-management-of-surgical-wound-complications-v2/file. Accessed Dec-2018.

Johnston SA, Tobias KM, eds. Veterinary Surgery: Small Animal Expert Consult, 2nd ed. St. Louis: Elsevier, 2018.

Kampf G, et al. Lack of sustained efficacy for alcohol-based surgical hand rubs containing 'residual active ingredients' according to EN 12791. J Hosp Infect. 2017;95:163-8.

Mathews KA, et al. A prospective study of intravenous catheter contamination. J Vet Emerg Crit Care. 1996;6(1):33-43.

National Healthcare Safety Network (NHSN). Chapter 9: Surgical site infection (SSI) event. In: NHSN Patient Safety Component Manual. 2019 Jan. Available at: https://www.cdc.gov/nhsn/pdfs/pscmanual/9pscssicurrent.pdf. Accessed Feb-2019.

Nicks BA, et al. Acute wound management: Revisiting the approach to assessment, irrigation, and closure considerations. Int J Emerg Med. 2010;3(4):399–407.

PennVet Instructional Technology. Donning a surgical gown & closed gloving. 2013. Available at: https://www.youtube.com/watch?v=gybhVnF3P4Q. Accessed Dec-2018.

Pittet D, et al. The World Health Organization Guidelines on Hand Hygiene in Health Care and their consensus recommendations. Infect Control Hosp Epidemiol. 2009;30(7):611–22.

Public Health Ontario (PHO). Provincial Infectious Diseases Advisory Committee (PIDAC). Best practices for cleaning, disinfection and sterilization of medical equipment/devices. 3rd ed. Toronto, ON: Queen's Printer for Ontario; 2013. Available at: https://www.publichealthontario.ca/en/eRepository/PIDAC_Cleaning_Disinfection_and_Sterilization_2013.pdf. Accessed Dec-2018.

Tanner J, et al. Surgical hand antisepsis to reduce surgical site infection. Cochrane Database Syst Rev. 2016;1:CD004288.

Verwilghen D, Singh A. Fighting surgical site infections in small animals. Vet Clin North Am Small Anim Pract. 2015; 45:243–76.

Widmer AF, et al. Surgical hand preparation: state-of-the-art. J Hosp Infect. 2010;74(2):112–22.

World Health Organization (WHO). Surgical hand preparation: state-of-the-art. In: WHO Guidelines on Hand Hygiene in Health Care: First Global Patient Safety Challenge Clean Care Is Safer Care. Geneva: World Health Organization; 2009. Available at: https://www.ncbi.nlm.nih.gov/books/NBK144036/. Accessed Dec-2018.

World Health Organization (WHO). Surgical Safety Checklist. 2009. Available at http://www.who.int/patientsafety/safesurgery/checklist/en/. Accessed Dec-2018.

World Health Organization (WHO). Appendix 10, Summary of the systematic review on surgical hand preparation. In: Global Guidelines for the Prevention of Surgical Site Infection. Geneva: World Health Organization; 2016. Available at: https://www.ncbi.nlm.nih.gov/books/NBK401149. Accessed Dec-2018.

World Health Organization (WHO). Protocol for surgical site infection surveillance with a focus on settings with limited resources. Geneva: World Health Organization; 2018. Available at: https://www.who.int/infection-prevention/tools/surgical/SSI-surveillance-protocol.pdf. Accessed Dec-2018.

Dental Procedures

Dental procedures often entail a significant risk of splash exposure to saliva, blood, and bacteria-laden debris (e.g. dental plaque). Procedures such as ultrasonic scaling can result in aerosolization of large numbers of bacteria. There is also potential for personnel to sustain cuts and abrasions from dental equipment or teeth during dental procedures. To reduce the risk of pathogen transmission from the animal's mouth to veterinary personnel during dental exams involving cleaning and other procedures that may aerosolize pathogens, the person performing the procedure and anyone in the immediate vicinity should wear:

- protective outerwear (e.g. designated lab coat, designated scrubs).
- protective eye glasses/goggles, or a full face shield.
- disposable gloves.
- surgical (i.e. nose and mouth) mask.

Dental procedures should be performed in a contained area away from other patients, personnel and high traffic areas. All surfaces in this area, including walls, floors and table, should be easy to clean and disinfect. Procedures such as bandage changes, wound care or placement of invasive devices (e.g. intravenous catheters, urinary catheters) should never be performed in close proximity to a dental procedure due to the risk of contamination by aerosolized bacteria.

Equipment disinfection

Like other surgical instruments, the recommended level of disinfection for dental instruments is based on the their Spaulding classification (see equipment disinfection section in Chapter: Cleaning, Disinfection, Sterilization). Many of the instruments used for these procedures are critical or semi-critical (e.g. periodontal scalers, mouth mirrors). Semi-critical equipment should undergo, at a minimum, high level disinfection between patients. Sterilization is required for critical items (RCDSO 2010 below). Most dental instruments can withstand steam sterilization (i.e. autoclave). For specialized equipment, or instruments with rotating components, follow the manufacturer's guidelines for cleaning and disinfection to prevent damage.

Antimicrobial prophylaxis

Antimicrobial prophylaxis is used to reduce the risk of disease from bacterial translocation during different procedures. While translocation likely occurs in a large percentage of dental procedures, the clinical consequences of this are typically very minor as disease rarely develop. In human dentistry, antimicrobial prophylaxis is reserved for a small subset of patients at particular risk of complications (Table 5 below) (Sarakiala-Kessel 2012 below). These are rare in veterinary medicine, and antimicrobial prophylaxis is likewise rarely indicated. When used, antimicrobial prophylaxis should begin before the procedure and typically not be continued after the procedure.

TABLE 5. Criteria for identification of veterinary patients that may require antimicrobial prophylaxis prior to dental procedures.

Patient Factors	Procedures
Patent ductus arteriosis	Dental cleaning that is expected to cause hemorrhage
Unrepaired cyanotic congenital heart disease	Any oral or periodontal surgery
Subaortic or aortic stenosis	Endodontic surgery
Previous infective endocarditis	
Imbedded pacemaker leads	

References

Royal College of Dental Surgeons of Ontario (RCDSO). Infection Prevention and Control in the Dental Office. Toronto ON: Royal College of Dental Surgeons of Ontario, 2010. Available at: https://az184419.vo.msecnd.net/rcdso/pdf/guidelines/RCDSO_Guidelines_Infection_Prevention_and_Control.pdf. Accessed Dec-2018.

Sarkiala-Kessel EM. Chapter 3: Use of antibiotics and antiseptics. In: Verstraete FJM, Lommer MJ (eds.) Oral and Maxillofacial Surgery in Dogs and Cats. Edinburgh: Saunders Elsevier, 2012;15-20.

Blood Donation

Transfusion of blood and blood products in veterinary practice is increasingly common. The limited number of centralized blood banks (particularly for cats) and time constraints on obtaining blood products from external sources have led many facilities to develop blood donation programs. These may involve resident animals that are kept on site for the purpose of blood donation (see Chapter: Non-Patient Animals) or use of local client or staff animals that are available as donors on short notice. These programs can be highly beneficial to patient care, yet carry some inherent risks of pathogen transmission.

There are two main infectious disease concerns with blood transfusions. One is transfusion of blood from an animal harbouring an infectious agent in its bloodstream. The second is bacterial or fungal contamination of blood products during collection, processing and storage. While rare, both can have potentially fatal consequences. Close attention to aseptic technique, processing of blood products in dedicated clean areas, use of standard operating procedures for handling blood, avoiding contamination of associated equipment and supplies, and careful screening of donors are critical to reduce these risks.

Donor screening

Donor screening helps to reduce transfusion-associated infections, but the risk can never be entirely eliminated because of limitations in test sensitivity/specificity, and the inability to test for all theoretically relevant microorganisms. Regardless, any facility that collects blood for transfusion must have clear protocols that take into account available recommendations and risks relevant to the geographic region. Some general recommendations include:

- Use standardized forms for enrollment and prior to each donation, including a consent form detailing potential risks to the donor.
- Do not use free-roaming cats as donors due to increased risk of infection with various pathogens (e.g. FIV, FeLV, FCV).
- Gather history and perform a physical exam prior to each donation, including:
  - temperature check
  - syndromic screening (e.g. presence or recent history of fever, depression, weakness, vomiting, diarrhea, coughing, sneezing)
  - health status of in-contact animals (e.g. housemates of donors)
  - inspection for external parasites
  - use of flea, tick and heartworm preventive practices
- Initially screen for relevant pathogens (see below), with periodic retesting at set intervals (e.g. every 6-12 months depending on pathogen, lifestyle and medical history).
  - Consider travel history when determining screening tests for individual animals.
- Keep records on all donors, recipients, type of transfusion, etc. to facilitate tracing should an adverse event occur or contamination of a product be detected.

Universal guidelines for disease screening for donors are impossible to develop because of regional differences in pathogen distribution. Economic factors may also play a role because the cost of screening may be significant. The appropriate frequency of testing is unclear and testing should be repeated whenever there have been potential new exposures. Periodic retesting is likely warranted but the appropriate time interval is unclear. Screening is recommended for pathogens that fulfill at least 3 of the following criteria (Wardrop 2016):

- documented to cause clinical infection by blood transfusion
- can be carried in the blood of clinically healthy animals
- can be detected in blood
- can produce disease that is severe or difficult to treat

For more information regarding recommended screening protocols and examples of intake questionnaires, see the 2016 ACVIM consensus statement on blood donor screening (Wardrop 2016).

Blood collection and processing

Blood must be collected using aseptic technique. Subsequent transfer and processing of blood should be performed in a clean laboratory area away from patients, areas where specimens such as fecal samples are handled and high traffic. Clear protocols must be in place regarding blood handling and processing practices. All blood samples should be clearly identified and logged, including the specific animal donor, person collecting the blood and date of collection.

Pre-transfusion evaluation

All blood products to be transfused should be given a close visual inspection for evidence of discolouration (an insensitive indicator of contamination but one that should result in further investigation of the product (e.g. culture)). The expiration date, donor species, product and blood type should also be verified prior to transfusion. Freezing a small aliquot (1 ml) of each transfused blood product is a good standard practice that allows for retrospective testing in the event of possible transmission-associated infection.

Screening for bacterial contamination of blood products

Routine screening of blood products for bacterial contamination is ideal; however, cost may be a limiting factor and it is unrealistic to expect that each unit of blood will be tested for an array of pathogens. Testing should be performed in any situation where contaminated blood products are suspected based on disease development in recipients, transfusion reactions, abnormal appearance of the blood or other factors. All samples from the same donor collected on the same day should be quarantined if testing is being performed on any one sample because of contamination concerns. If there is any suspicion that an infection attributable to a subclinically infected donor may have occurred, blood from that donor must be quarantined and tested or discarded, and the donor must be re-evaluated prior to any further donations.

Transfusion

Blood or blood products should be administered over a maximum of 4 hours and the administration set discarded after a single use (Day 2012 below).

Post-transfusion surveillance

Adverse transfusion events (e.g. fever, infection, anaphylaxis) should be logged by the clinic infection control practitioner so that the potential for bacterial contamination can be more readily investigated and identified should a cluster of potentially infectious adverse events be encountered.

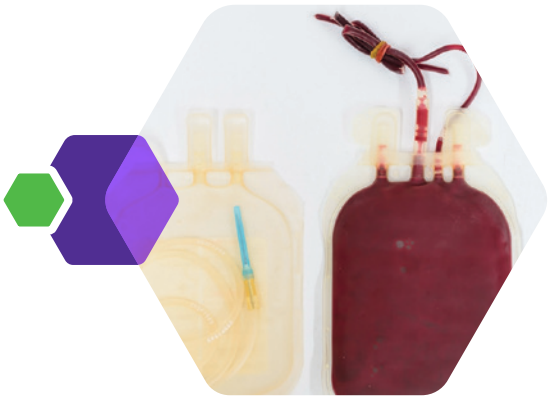
Passive surveillance of the health status of donors should also be performed by recording illnesses in donors within a short period of time (e.g. 48 hr) after donation. Good communication with owners is required when volunteer client or staff animals are used. If a donor develops clinical signs that could indicate a disease with transfusion-associated infection risk, blood products that remain from that transfusion should be quarantined until the cause of disease has been determined. In situations where blood or blood products have already been administered, testing of the donor may be important for identification of risks to the recipient. (See Chapter: Surveillance for more information regarding surveillance practices and programs.)

References

Wardrop KJ, et al. Canine and feline blood donor screening for infectious disease. J Vet Intern Med. 2005;19:135-42.

Wardrop KJ, et al. ACVIM Consensus Statement: Update on canine and feline blood donor screening for blood-borne pathogens. J Vet Intern Med. 2016;30:15-35.

Day MJ, Kohn B (eds). BSAVA manual of canine and feline haematology and transfusion medicine. 2nd ed. Gloucester UK: British Small Animal Veterinary Association; 2012.



Rehabilitation and Physical Therapy

Rehabilitation is an increasingly popular field of veterinary medicine and raises some unique infection control issues. Rehabilitation is often used in patients that are recovering from an infection (e.g. surgical site infection), carrying multidrug-resistant pathogens (e.g. due to prolonged antimicrobial use and hospital exposure) and susceptible to infection from immobility due to neurological or orthopedic problems (e.g. paralysis or paresis leading to pressure sores, or urinary dysfunction increasing the risk of urinary tract infection). Cancer patients undergoing rehabilitation may be further immunosuppressed due to chemotherapy or radiation therapy. Therefore, this population should be considered at increased risk of both being infectious and susceptible to infection.

Virtually no research is available pertaining to infectious disease risks or infection control practices associated specifically with rehabilitation in veterinary medicine, but basic infection prevention and control principles still apply. These include hand hygiene, cleaning and disinfection of the environment and equipment surfaces, proper patient handling to reduce pathogen transmission between patients and hospital personnel, and management of infectious syndromes. Details for each of these practices can be found in the corresponding chapters of these guidelines.

Patient assessment

Patient evaluation and development of a rehabilitation plan is a standard practice, and should include consideration of infectious disease issues, such as patient susceptibility to infection and their likelihood of shedding an infectious agent (e.g. ongoing/previous infection, presence of open wounds, antimicrobial therapy). The rehabilitation plan should be modified as necessary if the patient's status changes (e.g. planned activities with water vs no-water)

If a patient is potentially infectious, and rehabilitation cannot be postponed, additional measures should be taken such as treating the animal at the end of the day to limit the risk of exposure to other patients and to allow for more thorough cleaning and disinfection after the session, use of enhanced barriers (e.g. gloves, additional protective outerwear), and extra attention to hand hygiene.

Hydrotherapy

Hydrotherapy (the use of a pool or tank with or without an underwater treadmill) is a common rehabilitation tool. While formal research studies are lacking, anecdotal evidence suggests that the risk of hydrotherapy-associated infection is limited. However, there are theoretical concerns, particularly transmission of multidrug-resistant pathogens such as *Pseudomonas spp*, methicillin-resistant *Staphylococcus pseudintermedius* (MRSP) and enteropathogens.

While hydrotherapy is likely low-risk overall, certain precautions should still be used to reduce risks to patients and the facility as a whole. On each treatment occasion, the patient's risk status should be reassessed and changes made as necessary (see patient assessment section above). Prior to entering a pool or treadmill, the patient should be examined to ensure there are no new open wounds or skin lesions. Patients whose haircoats are visibly dirty should be brushed, rinsed or washed prior to use of hydrotherapy equipment, depending on the amount and nature of contamination.

A hydrotherapy log should be kept of the patient, personnel, date and time of each use of the pool/tank in the event that any contact tracing is required.

Microbiological testing of hydrotherapy water

Routine testing of municipal water at the source or the pool/tank water is not indicated. Water testing in these cases is typically only used as part of an epidemiologic investigation in response to an outbreak where hydrotherapy water is considered a possible source. Facilities using wells or other non-municipal sources (e.g. cisterns) that are not otherwise regularly tested like municipal water sources should ensure that microbiological testing is performed as per local recommendations.

Pool maintenance

For complete maintenance requirements, refer to manufacturer guidelines. General recommendations include:

- Keep the pool area and filters clean, removing hair and debris daily or more often as required.
- Monitor and maintain water quality, disinfectant level and water chemistry balance, to avoid persistent contamination as pool water is reused, and to prevent skin and eye irritation for patients and staff in contact with the water.
  - Check water chemistry levels at least daily to make corrections as indicated. Record all measurements to facilitate compliance and identify problems with quality or monitoring.
  - Establish written protocols that detail water testing, routine water treatment and the response to any abnormalities.

While chlorine is the most commonly used pool disinfectant, bromine (another halogen) is also widely available. Bromine can be an effective water disinfectant at 3-6 ppm, although there is less information regarding effective times and concentrations for management of fecal contamination. Bromine activity is less dependent on pH than chlorine, but standard water quality monitoring practices still need to be followed.

Various other water treatment systems are available, including ozone and ultraviolet light; however, they are probably unnecessary in a well-managed facility. They may provide additional benefits in facilities with high traffic or inadequate source water quality, but they should not be used in place of good management.

Establish written protocols that detail water testing, routine water treatment and the response to any abnormalities (e.g. defecation in the water) for hydrotherapy pools.

Fecal contamination of pools

If a patient defecates in the water:

- Remove the animal immediately and return it to its cage/kennel.
- Promptly wash hands.
- If staff were in the water with the animal, showering is recommended.
- Remove as much of the fecal material from the pool as possible using a net or bucket, taking care to prevent contamination of personnel or the environment in the process. This should be done as quickly and gently as possible to avoid breaking up pieces of solid waste and further dispersing the contamination.
- Do not use the pool vacuum to remove gross contamination unless water can be directly discarded using a “waste” setting (or equivalent) that bypasses the pool filter.
- Initiate a water treatment plan once gross contamination has been removed.
- If gross contamination cannot be adequately removed, drain all the water, clean the pool surfaces of any residual debris, refill and treat the water as follows: maintain fresh water at 2 ppm chlorine, pH of 7.5 or less and temperature of 25°C (77°F) or higher for 25 minutes prior to next use.
- Change or clean and disinfect the filter after treatment.
- The pool can be used once water parameters have returned to normal.
- Log the incident, including patient identifier and the cleaning and disinfection measures that were used.
- Bathe the patient, especially before immersion in the clean pool or contact with other patients.

A more aggressive approach using higher chlorine levels and longer contact times is justified if a dog passes diarrhea in the pool, which is more difficult to remove and more likely to contain environmentally tolerant enteropathogens.

Underwater treadmill maintenance

Underwater treadmills should be drained between uses, therefore water quality is less of a concern than treadmill cleaning, surface disinfection and maintenance. Holding tanks and reservoirs are commonly used to store the water from underwater treadmills. To reduce the risk of pathogen accumulation or proliferation, the water should be treated and managed as for a pool.

After draining, the treadmill should be inspected for and cleared of any gross contamination. Regular use of an appropriate disinfectant on tolerant surfaces is prudent; however, the appropriate frequency of disinfection is unknown. Cleaning, disinfection and complete drying of the interior surfaces and any exterior surfaces in contact with an animal or human should be performed if the patient is known or suspected to be shedding an infectious pathogen.

If fecal contamination occurs, management is the same as for fecal contamination of pools.

Personnel in pools

Handlers accompanying animals into pools should be healthy with no skin lesions. Water clothing worn in the pool should not be taken home but should be laundered in the clinic or by a commercial laundry service (see Chapter: Laundry and Waste Management).

Dry rehabilitation therapy

Items such as mats, stairs, fitness balls, balance boards, small jumps (cavalettis), carts and dry treadmills may be used during dry land rehabilitation therapy. These items should be amenable to cleaning and disinfection (e.g. impermeable) and should be inspected regularly to identify surface defects that might facilitate pathogen survival. The required level of cleaning and disinfection should be determined for each item based on the Spaulding classification (see equipment disinfection section in Chapter: Cleaning, Disinfection, Sterilization), and recorded in the clinic infection control manual for easy reference. Fabric items such as reusable elastic bandages, slings and braces should be laundered in the clinic and hot-air dried (see Chapter: Laundry and Waste Management).

References

Centers for Disease Control and Prevention (CDC). Water use in hydrotherapy tanks. 2016. Available at: http://www.cdc.gov/healthywater/other/medical/hydrotherapy.html, Accessed Dec-2018.

Centers for Disease Control and Prevention (CDC). Fecal incident response recommendations for aquatic staff. 2018. https://www.cdc.gov/healthywater/swimming/pdf/fecal-incident-response-guidelines.pdf. Accessed Dec-2018.

Craun GF, et al. Outbreaks associated with recreational water in the United States. Int J Environ Health Res. 2005;15:243-62.

Davis B. Whirlpool operation and the prevention of infection. Infect Control. 1985:394-7.

De Jonckheere JF. Hospital hydrotherapy pools treated with ultra violet light: bad bacteriological quality and presence of thermophilic Naegleria. J Hygiene. 1982;88:205-14.

Edlich RF, et al. Water treatment of hydrotherapy exercise pools. J Burn Care Rehab. 1988;9:510-5.

Insler MS, Gore H. Pseudomonas keratitis and folliculitis from whirlpool exposure. Am J Ophthalmol. 1986;101:41-3.

Jacobson J. Pool-associated Pseudomonas aeruginosa dermatitis and other bathing-associated infections. Infect Control. 1985:398-401.

Laarhoven LM, et al. Longitudinal study on methicillin-resistant Staphylococcus pseudintermedius in households. PLoS ONE. 2011;6:e27788.

Meldrum R. Survey of Staphylococcus aureus contamination in a hospital's spa and hydrotherapy pools. Commun Dis Public Health. 2001;4:205-8.

Podewils LJ, et al. Outbreak of norovirus illness associated with a swimming pool. Epidemiol Infect. 2007;135:827-33.

Polgreen PM, et al. A statewide outbreak of Cryptosporidium and its association with the distribution of public swimming pools. Epidemiol Infect. 2012;140:1439-45.

Schiemann D. Experiences with bacteriological monitoring of pool water. Infect Control. 1985:413-417.

Sehulster L, et al. Guidelines for environmental infection control in health-care facilities. Recommendations of CDC and the Healthcare Infection Control Practices Advisory Committee (HICPAC). MMWR Recomm Rep. 2003;52(RR-10):1-42.

Additional Considerations

Safety of Clinic Personnel

Good infection prevention and control strategies in all areas of the veterinary clinic will enhance the safety of clinic personnel. This includes, but is not limited to, maintaining good hand hygiene, using appropriate personal protective equipment (PPE), cleaning and disinfection methods, and following appropriate procedures for patient management and care. Details of these practices can be found throughout this document. This section highlights some other important considerations for maintaining the safety of clinic personnel as they relate to infection prevention and control. Employers and hospital managers should be aware of additional occupational health risks, and abide by workplace safety regulations in their region.

Bites and scratches

Bites and scratches are an inherent risk in veterinary medicine and a common cause of occupational injury and illness. From January 2007 to June 2011, injury from animal contact accounted for 82% of workers' compensation claims submitted to the American Veterinary Medical Association Professional Liability Insurance Trust (JAVMA News 2012). In a survey of US veterinarians, approximately two-thirds had sustained a major animal-related injury at some time, and bites and scratches accounted for just over one-third of these injuries (Landercasper 1988). Approximately 3% to 18% of dog bites and 28% to 80% of cat bites become infected (Davies 2000). Most dog and cat bite wound infections are caused by a mixture of aerobic and anaerobic bacteria (Talan 1999). It should be assumed that all animals are carrying potentially zoonotic pathogens in their mouths.

In general, veterinary personnel should be able to recognize behaviours indicative of fear, anxiety or stress in animals, and situations that are associated with an increased tendency for an animal to bite. Bite prevention practices must be guided by professional judgment and use of accepted methods to reduce fear, anxiety and stress in patients. Personnel should take all necessary precautions to prevent animal-related injuries in the clinic. These may include physical restraint or chemical restraint (sedation or anesthesia) of an animal. Techniques for reducing potential injurious behavior should be use whenever possible (fearfreepets.com), but appropriate equipment (e.g. different sizes of muzzles, bite-resistant gloves, catch pole, cat bags) should be readily available if necessary. Such equipment should also be as easy to clean as possible. Experienced veterinary personnel rather than owners should restrain animals for procedures whenever possible. Personnel must always be aware of changes in their patients' behaviour which may precede attempts to bite. Veterinary personnel should not let client perceptions or attitudes prevent them from using appropriate bite-prevention measures (e.g. muzzling). Notes should be made in the files of patients that routinely exhibit fearful or aggressive behaviour to alert staff.

If anyone is bitten by an animal:

- Immediately wash the wound thoroughly with plenty of soap and water, and remove any visible debris
- Report bite incidents to local public health (where required by law)
- Medical attention is particularly important for any bite that:
  - is on a hand or over a joint
  - is over a tendon sheath, such as bite on the wrist or the ankle
  - is on the face
  - is in the genital area
  - is over a prosthetic device or implant
  - is over an area with pre-existing chronic swelling (edema)
  - causes a large amount of tissue damage (e.g. a deep tear or tissue flap)
  - is sustained by a person without a functional spleen
  - is sustained by a person with an immune system compromised by disease or drug therapy

Regardless of the location of the wound, if the area becomes increasingly painful or swollen, if the wound develops a discharge, or if the person develops a fever or swollen lymph nodes, consult a physician as soon as possible.

Scratches pose a lower risk of infection compared to bites, but can still cause significant tissue damage that is then susceptible to infection by endogenous or exogenous bacteria. The risk of rabies transmission from scratches is negligible, unless the wound is also contaminated with saliva from the animal. Nonetheless, as for bite wounds, scratch wounds should be washed immediately with plenty of soap and water, and any debris removed. Subsequent wound care to prevent infection depends on the nature and site of the wound. If in doubt, or if the victim has a compromised immune system for any reason (see above), it is best to seek medical attention.

A physician will decide (in some cases in consultation with public health personnel) if antimicrobial therapy, tetanus vaccination, rabies post exposure prophylaxis, or any additional treatment (e.g. lavage, debridement, sutures) are necessary for bite or scratch wounds. Most bite wounds are not sutured in order to promote drainage and reduce the risk of infection.

Emergency contact information (i.e. physician, local public health department) should be clearly posted in the clinic.

All bites or scratches should be reported to the clinic infection control practitioner (ICP) and the injury documented. A pre-determined bite protocol that reflects applicable labour laws helps ensure that all necessary reporting and documentation occur. Bites and scratches should not be considered "part of the job" and summarily dismissed. Even seemingly small, innocuous injuries can develop severe complications. (See Table 2 in Chapter: Personal Protective Equipment for infectious agents related to bites and scratches.)

Regular review of injuries (including those unrelated to infectious disease risks) is useful to identify trends in behaviour or procedures that may be associated with injuries and to develop protocols to reduce the risk of injuries. Documentation is also important for employees in the event that serious health problems subsequently develop.

Sharps

Injuries from needles and other sharp implements are common in veterinary medicine but are largely preventable. Although there is not the level of risk of bloodborne pathogen exposure in veterinary practice as there is in human medicine, serious outcomes can result following needlestick or other sharps injuries. These may include physical trauma, secondary infection and drug reactions due to live vaccines and other substances (i.e. toxic, allergic, idiosyncratic).

Proper sharps handling

Proper sharps handling practices are a practical yet effective way of reducing workplace injuries in veterinary clinics. Use appropriate barriers (e.g. closed toed shoes) and safe work practices when using sharp instruments and devices (e.g. needles, scalpels, etc.), after procedures and when cleaning used instruments.

To prevent needlestick and other sharps injuries:

- Never remove needle caps by mouth.
- Do not bend or manipulate needles in any way.
- Do not pass uncapped needles to another person.
- Ensure proper animal restraint to reduce inadvertent needlestick injuries from animal movement.
- Do not recap needles by hand. If recapping is required, use the "one-handed scoop" technique (see below), forceps or a needle cap holder.
- **Ensure that approved point-of-use sharps disposal containers are located everywhere needles are handled.** These containers are puncture-resistant, leak-proof, and prevent removal (both accidental and intentional) of discarded sharps.
- Always dispose of sharps immediately in an approved sharps disposal container. Do not overfill disposal containers.
- **Never dispose of needles or other sharps into anything other than an approved sharps container**, even if they are capped or otherwise contained. This reduces the risk of accidental injury to veterinary personnel, patients, clients and non-veterinary personnel (e.g. waste disposal personnel).
- Ensure all veterinary personnel have received training on proper sharps handling.

The most important precaution for preventing needle-stick injuries is to **avoid recapping needles**. Recapping needles causes more injuries than it prevents. When it is absolutely necessary to recap needles as part of a medical procedure or protocol:

- Use a mechanical device such as forceps or hemostats to replace the cap on the needle.
- Alternatively, the needle can be recapped using the **"one-handed scoop"** technique:
  - Place the cap on a flat horizontal surface.
  - Holding the syringe with the attached needle, or the needle hub alone (when unattached), scoop up the cap with the needle by sliding the needle tip inside, without touching the cap with one's other hand.
  - Once the point of the needle is covered, tighten the cap by pushing it against an object, or by pulling the base of the needle cap onto the hub of the needle with the same hand holding the syringe.

After injecting live vaccines or aspirating body fluids or tissue, the used syringe should be placed in a sharps container with the needle attached. Following most other veterinary procedures, the needle and syringe may be separated for disposal of the needle in the sharps container. This is most safely accomplished by using the needle removal device on an approved sharps container, which allows the needle to drop directly into the container without being handled or touched.

As with bites, needlestick injuries should be documented and, if necessary, investigated. Requiring documentation of needlestick injuries can help detect a change in rates, allow for detection of factors that are recurrently associated with injury, and help develop a plan to reduce injuries.

Recapping needles causes more injuries than it prevents.

Sharps safety for clients

Periodically, owners may be required to treat their animals at home with injectable medications (i.e. insulin, subcutaneous fluids). In these situations, it is the responsibility of the attending veterinarian to:

- Provide (and document) training on how to handle sharps, including injection and disposal practices.
- Provide an approved sharps container or give clients clear instructions regarding how to obtain one.
- Ensure that the client is able to safely handle and dispose of sharps.
- Advise clients that the sharps container should be returned to the clinic for disposal when 3/4 full, and exchanged for a new container (if necessary).

Used sharps are considered biomedical waste in veterinary practices. Dispose of used sharps containers in accordance with regulations from municipal and/or provincial/territorial authorities.

Clinic laboratory

Activities involving the clinic lab may include fecal parasitological testing, hematological testing, urinalysis, cytological analysis of other samples, as well as more intensive procedures such as bacterial and fungal culture. Any activities involving biological specimens pose some degree of risk of exposure of personnel and contamination of the environment. The risk varies, and may range from quite low (e.g. hematology) to high (e.g. bacterial and fungal culture). When pathogens are handled, especially when they are cultivated, clinics must ensure that they are compliant with all relevant regulations regarding these activities.

Laboratory personnel

All personnel who handle specimens should be trained in specimen handling, testing and biosafety practices. The training should specifically relate to their activities, as the practices and risks associated with some (e.g. use of an automated blood analyser) can be very different from others (e.g. fungal culture). Clear guidelines for these practices, required personal protective equipment, cleaning and disinfection, waste disposal and spill response (see below) are required. Training, as always, needs to be documented.

Laboratory area

A designated area of the clinic should be used for specimen processing, even if it only involves processing samples to send to an external diagnostic laboratory. This should be separate from treatment and surgery rooms so as to decrease the risk of contamination of these areas. After processing a specimen:

- Dispose of sharps such as microscope slides and glass pipettes in approved sharps containers.
- Dispose of samples from animals with suspected or known infectious diseases as infectious waste.
  - Regulations vary by region and facilities must be compliant with legal requirements for biohazard/infectious disposal in their area.
- Clean and disinfect specimen processing areas immediately after use.
- Perform hand hygiene.

Handling of biological specimens

Urine from animals with suspected urinary tract disease, and all feces, aspirates, and swabs should be treated as potentially infectious material, even though they are not necessarily considered biomedical waste. Proper handling includes:

- Wearing **protective outerwear** (e.g. lab coat) and **disposable gloves** when handling specimens.
  - Discard gloves and wash hands immediately after handling these items.
  - Avoid touching clean items (e.g., microscopes, telephones, pens) while handling specimens or prior to performing hand hygiene.
- Carefully filling specimen containers so as to p**revent contamination of outer surfaces** with feces, blood or other materials.
  - If contamination occurs, clean and disinfect the surface with an appropriate product.
- Appropriate packaging of biological specimens that are being sent to external laboratories.
  - Store samples in **leak-proof plastic containers** designed for the specimen (e.g. blood vials, urine containers, fecal cups) and then place them in a secondary clean, sealed plastic bag prior to shipping.
  - If samples must be refrigerated, do not store them in refrigerators that also contain medications, vaccines or food (human or animal).

Spill plan

Even with good storage and handling practices, spills of biohazardous materials may occur. A written spill plan should be available that details the response to large and small spills, which should be appropriate for the substances that are handled. In general, to respond to a spill:

1. Notify people in the area (or that are likely to enter the area during the spill cleanup process) that a spill has occurred.
2. Wear proper protective outerwear (e.g. laboratory coat, gloves).
3. Contain the spill as quickly as possible to prevent further spread, if necessary.
4. Gather all required materials (e.g. paper towels, disinfectant, biohazard bag) prior to beginning clean up.
5. Remove as much gross material as possible (including solids and liquids) using absorbent materials such as paper towels and place them directly into a biohazard bag.
   - Broken glass must be placed in a sharps disposal container or other rigid container. Do not place directly in a biohazard bag.
6. Apply a disinfectant that is appropriate for the pathogens that may be present to the contaminated area. Ensure the surfaces remain wet for the indicated contact time for the product.
7. Wipe the area and dispose of paper towels in the biohazard bag.
8. Remove protective clothing and perform hand hygiene.
9. Consider why the spill happened and whether similar spills can be prevented in the future.

In-house bacterial and fungal cultures

Any facility performing bacterial or fungal culture must consider the increased risks associated with these procedures, as well as regional regulations for manipulating biohazardous agents. Some countries (including Canada) also have strict federal regulations in this regard. These mandate full compliance with biosafety level 2 (BSL-2, also referred to as containment

level 2, CL2) practices for any culture of potential pathogens. Regardless of regulations, any clinic performing bacterial culture should be knowledgeable of standard laboratory biosafety practices and be compliant with BSL-2 practices. Guidance for appropriate laboratory safety practices can found on a number of websites, including those listed in Table 1 below.

Compliance with these practices is feasible for clinics with adequate infrastructure and staffing, but proper containment may not be feasible in terms of cost or time requirement for others. If culture is performed, it must be done right, in terms of both quality and safety. While uncommon in most areas, there are situations where BSL-3 pathogens could be encountered and cultured with routine culture practices. Some examples include *Yersinia pestis* (plague), *Francisella tularensis* (tularemia) and *Brucella* spp. Cultures should never be performed in an on-site clinic laboratory on specimens from animals suspected of harbouring BSL-3 pathogens.

Parasitological examination

Fecal parasitological testing is very commonly performed, and fecal contamination of the local environment is common. Such testing should be performed in a dedicated laboratory area and on surfaces that are easily cleaned and disinfected. Sinks used for fecal analysis should not be used for non-laboratory activities such as cleaning patient equipment or other items.

TABLE 1. Laboratory biosafety resources

Resource	Link
Public Health Agency of Canada Laboratory Biosafety Guidelines, Standards, and Handbook	https://www.canada.ca/en/public-health/services/canadian-biosafety-standards-guidelines.html
Public Health Agency of Canada Veterinary Practices: Physical Design and Operational Practices for Diagnostic Activities	https://www.canada.ca/content/dam/phac-aspc/documents/services/canadian-biosafety-standards-guidelines/guidance/veterinary-practices-physical-design-operational-practices-diagnostic-activities/pub-eng.pdf
US Centers for Disease Control and Prevention Biosafety in Microbiological and Biomedical Laboratories	http://www.cdc.gov/biosafety/publications/bmbl5/
World Health Organization Laboratory Biosafety Manual	www.who.int/csr/resources/publications/biosafety/Biosafety7.pdf

Necropsies

Necropsies are high risk procedures because of potential contact with infectious body fluids, aerosols, and contaminated sharps. Non-essential persons should not be present during necropsy procedures in order to minimize exposure of personnel to these hazards. Personnel involved in or present at necropsies should wear:

- protective outerwear (e.g. designated lab coat, designated scrubs).
- disposable gloves.
- protective eye glasses/goggles, or a full face shield.

In addition, when opening the body cavities of larger animals or for any other heavy cutting, cut-proof gloves which can be washed in the laundry should be used to prevent accidental injury from necropsy blades. Additional precautions for respiratory protection (including environmental controls and face masks) should be employed if power equipment is used, since these instruments increase the amount of potentially infected material that becomes aerosolized. Precautions should also be taken to contain all body fluids from the animal, particularly if release of a large volume is anticipated.

It is recommended that in-clinic necropsies not be conducted on any animal suspected of being infected with a pathogen requiring biosafety precautions above containment level 2 (e.g. *Chlamydophila psittaci, Coxiella burnetti, Francisella tularensis).* Instead, the entire body should be submitted to an approved diagnostic laboratory. Ensure that all requirements for shipment of biological samples are met (these can usually be provided by the laboratory in question), including providing notification

of any suspected infectious disease in order to protect laboratory personnel. Material Safety Data Sheets (MSDS) for human pathogens, including many zoonotic pathogens, are available on the Public Health Agency of Canada (PHAC) website (see References). These sheets list the recommended precautions for handling these pathogens and potentially infectious materials as safely as possible. For more information on risk group classification of infectious agents, visit the American Biological Safety Association website (see References). Information on the requirements for the different containment levels needed to handle infectious pathogens can be found in the Canadian Biosafety Standards, which are also available online (see References).

Vaccination of personnel

Vaccination should be considered a last line of protection, but is important for certain diseases. Decisions regarding vaccination policies for personnel should consider the risk of exposure, the severity of disease, whether the disease is treatable, the transmissibility of disease, as well as the quality and safety of the vaccine.

Rabies: Rabies vaccination is indicated for anyone who has a greater than average risk of exposure to the virus. **All veterinary personnel that might have contact with animals should therefore be vaccinated against rabies**, except in areas that have been formally declared rabies-free (e.g. some islands or specific countries). This includes lay staff who might have periodic animal contact, such as front office staff. Even animals that are kept indoors can be exposed to rabies by bats, and the disease may not be suspected on initial admission. Rabies vaccines for humans are generally considered safe and highly effective. In areas where rabies is endemic, rabies titres should be checked every 2 years to ensure that the minimum accepted titre is maintained, with re-vaccination provided as required. Additional information on rabies vaccination in people is available in the Canadian Immunization Guide (2015) and on the Centers for Disease Control and Prevention (CDC) rabies website (see References).

Tetanus: Although bites and scratches are very low risk for tetanus infection, cuts and scratches from other objects or soil contamination of puncture wounds are still a risk. Therefore, tetanus vaccination is indicated in veterinary personnel. Boosters are generally administered every 10 years.

Influenza: Human influenza is a common and highly transmissible disease, even though it is generally considered minimally transmissible to companion animals (but particularly to ferrets and pet pigs). Nonetheless, infected veterinary personnel can rapidly infect their colleagues, and veterinary clinics can act as sources of community spread if infected employees are present. It is reasonable for veterinary clinics to recommend annual influenza vaccination of all personnel (as per the recommendations of the Canadian National Advisory Committee on Immunization (NACI)), and to ensure that personnel have time to visit their physician or a vaccination clinic for this purpose. Employees should also be encouraged to stay home if they are ill.

High-risk personnel

Strict adherence to routine infection control practices should protect against most potential infectious disease risks. High-risk individuals should consult with their physician (and other medical personnel as required) to identify potential hazardous situations that should be avoided or handled using altered practices or precautions. Potential high-risk situations that might be encountered (e.g. managing leptospirosis suspects) should be considered in advance in order to develop a plan to reduce the risk of exposure. It is also important to remember that the **infection control practices of the entire clinic team play a critical role in the overall safety of its individual members.**

Pregnant personnel

Pregnant women working in a veterinary clinic setting may encounter pathogens that can cause disease in any individual but also subsequently risk fetal infection or death (e.g. Salmonella, Leptospira spp, Brucella spp, Coxiella burnettii), as well as pathogens that rarely cause disease in pregnant or non-pregnant individuals but which can cause devastating disease in a developing fetus (e.g. Toxoplasma gondii). There is also a decrease in cell-mediated immunity in the third trimester that may increase the risk of certain common infectious diseases, particularly those caused by viruses and fungi.

All personnel, including temporary lay personnel, kennel staff, students and volunteers, should receive education and training about injury prevention and infection control.

Immunocompromised individuals

In general, immunocompromised individuals are at increased risk of infection from pathogens that do not (or rarely) cause disease in healthy individuals, and severe disease compared to infections in immunocompetent individuals (e.g. salmonellosis, cryptosporidiosis). Individuals with a known immunodeficiency should take extra care to follow routine infection prevention practices and avoid high risk situations.

Training and education of personnel

Personnel training and education are essential components of an effective infection control program. All personnel, including temporary lay personnel, kennel staff, students and volunteers, should receive education and training about injury prevention and infection control during their initial orientation and periodically thereafter. Additional training should be provided as recommendations change or if problems with infection control practices are identified. Training should emphasize awareness of the hazards associated with individual work duties, and prevention of zoonotic disease exposure. Staff participation in training should be documented by the infection control practitioner (ICP). A variety of helpful training resources are available (Gibbins 2015 below).

References

American Biological Safety Association. Risk group database. Available at: https://my.absa.org/Riskgroups. Accessed Dec-2018.

Center for Disease Control and Prevention (CDC). Rabies – Medical care – Rabies vaccine (updated 24-Sep-2014). Available at: https://www.cdc.gov/rabies/medical_care/vaccine.html. Accessed Dec-2018.

Davies HD. When your best friend bites: A note on dog and cat bites. Can J Infect Dis. 2000;11(5):227-9. PubMed PMID: 18159293.

Fear Free. https://fearfreepets.com/fear-free-research/. Accessed Dec-2018.

Gibbins JD, MacMahon K. Workplace safety and health for the veterinary health care team. Vet Clin North Am Small Anim Pract. 2015;45:409-426

Harding AL, Byers B. Epidemiology of laboratory-associated infections. In: Fleming DO, Hunt DL, eds. Biological safety: principles and practices. Washington, DC: ASM Press, 2000;35-54.

JAVMA News. AVMA PLIT lists top claims for business insurance. 15-Oct-2012. Available at: https://www.avma.org/News/JAVMANews/Pages/121015m.aspx. Accessed Dec-2018.

Landercasper J, et al. Trauma and the veterinarian. J Trauma. 1988;28(8):1255-9. PubMed PMID: 3411647.

Public Health Agency of Canada (PHAC). Canadian Biosafety Standard (CBS). 2nd ed. 2015. Available at: https://www.canada.ca/en/public-health/services/canadian-biosafety-standards-guidelines/second-edition.html. Accessed Dec-2018.

Public Health Agency of Canada (PHAC). Pathogen safety data sheets (updated 24-Jul-2018). Available at: http://www.phac-aspc.gc.ca/msds-ftss/index-eng.php. Accessed Dec-2018.

Public Health Agency of Canada (PHAC). Rabies vaccine (updated Jan-2015). In: Canadian immunization guide: Part 4 - Active vaccines. Available at: https://www.canada.ca/en/public-health/services/publications/healthy-living/canadian-immunization-guide-part-4-active-vaccines/page-18-rabies-vaccine.html. Accessed Dec-2018.

Talan DA, et al. Bacteriologic analysis of infected dog and cat bites. N Engl J Med.1999;340(2):85-92. PubMed PMID: 9887159.

Weese JS, Prescott JF. Assessment of laboratory and biosafety practices associated with bacterial culture in veterinary clinics. J Am Vet Med Assoc. 2009;234:352-35

Client Visitation

Given the strong bond between owners and their pets, it is understandable when clients wish to visit their hospitalized pets. However, animals carrying transmissible pathogens pose a potential risk to other animals at the clinic and at the owners' home, as well as to the owners themselves, other household members and clinic employees. As a policy, clients should not be allowed to visit animals that are considered potentially infectious. Under extenuating circumstances, such as an animal whose condition is imminently life-threatening or where euthanasia decisions are being made, it is reasonable to accommodate controlled visitation when risks can be identified and measures can be taken to reduce those risks. The risks posed by some pathogens such as rabies virus or Yersinia pestis (the cause of plague) are such that visitation should not be permitted. Therefore, visitation should be considered on a case-by-case basis evaluating the pathogens of concern, the ability to implement infection control measures, and the understanding and acceptance of any risks by the owner. Consider additional limitations for visitation by children (especially young children), and ensure all visitors are escorted and supervised by clinic personnel in order to ensure control measures are followed. Hand hygiene should emphasized in all cases, and enhanced infection control practices such as personal protective equipment may be required as well. Clinics should be aware of the liability associated with client visitation, especially when children are involved. In some scenarios it may be easier to manage the animal and the visitors in a designated exam room, in which case protocols must be followed for movement of infectious patients within the clinic, as well as thorough cleaning and disinfection of the room after the visit.

As a policy, clients should not be allowed to visit animals that are considered potentially infectious.

Non-Patient Animals

In some veterinary hospitals, a variety of non-patient animals may be present, including boarders, recurrent day care animals, pets that accompany staff to work, blood donor animals and resident (clinic) animals. Their presence poses some risk to both the non-patient animals and patients alike, and though the degree of risk has been minimally investigated (Ghosh 2012), a facility-specific assessment should be made of the benefits. Transmission of pathogens such as canine influenza via transient non-patient animals has been identified (Weese, 2019), highlighting the potential risks of having non-patient animals present in the clinic.

From an infection control standpoint, non-patient animals should not be present in veterinary hospitals. However, if it cannot be avoided there are measures that can and should be taken to reduce the risk of pathogen transmission between non-patient animals and patients or staff.

From an infection control standpoint, non-patient animals should not be present in veterinary hospitals.

Boarding / day care

Transmission of pathogens and disease outbreaks amongst boarding/day care animals are poorly described in the veterinary literature. Outbreaks of canine upper respiratory disease complex, undifferentiated diarrhea, ringworm and canine papillomatosis (Lane 2017) have occurred in day cares. **Veterinary hospitals considering boarding/day care should assess whether it can be done in a manner that does not pose undue risk to hospitalized patients or boarding animals.** Some of the factors to consider are listed in Table 2.

Although boarding animals may appear clinically normal, there is always a risk of subclinical shedding of infectious pathogens which can be spread through direct or indirect contact. Creating more crowded conditions can also increase the risk of pathogen transmission simply by the relative proximity of a larger number of animals.

If boarding is permitted, careful consideration must be given to selection of boarders and boarding practices:

- Insist that boarding animals be on a regular preventative health program that includes a full physical examination by a veterinarian at least annually, regular vaccination, and internal/external parasite control, as appropriate.
  - Ideally boarders are patients at the same veterinary clinic so their medical history is readily available. If not, a copy of the medical record from the clinic where the animal was last examined should be provided.
- Avoid introduction of high-risk animals (e.g. recently infectious, from a shelter, or imported) into the facility.
- Boarders should be up-to-date on all core vaccinations for a given geographic area (Ford 2017), as well as *Bordetella bronchiseptica* and canine parainfluenza virus (kennel cough) for dogs. Canine influenza vaccination is also prudent in areas where the virus is circulating.
  - Time vaccinations so that there is a reasonable expectation of protective immunity at admission.
- Perform a behavioural assessment to determine how the animal is likely to respond to the presence of other animals of the same or different species, as well as any behavioural triggers that need to be avoided. Note this in the boarder's file in some way.
- Outline infection control measures to clients before initial admission to facilitate compliance and reduce the potential for conflict if an animal must be excluded.
- Use daily syndromic surveillance to detect clinical issues (see Chapter: Surveillance). These syndromes do not always indicate that an infectious disease is present, but they indicate the need for a veterinary examination to determine whether boarding of the animal poses any additional risk. Relevant syndromes to screen for include, but are not limited to:
  - lethargy
  - vomiting
  - diarrhea
  - coughing
  - sneezing
  - presence of external parasites
  - oral papillomas
  - skin lesions

- Restrict direct and indirect contact between boarders, and between boarders and clinical cases. Complete physical and procedural separation is ideal, including separate food bowls, water bowls and toys.
  - If group contact is unavoidable, keep specific groups of animals together. This helps limit any disease events that may occur to a smaller group, and makes interventions (e.g. isolation, temporary exclusion) more practical to implement.
- Have a clear, written protocol manual that details all activities involved with the day care and all infection control practices that are undertaken.

TABLE 2. Important considerations when determining whether boarding should be permitted at a veterinary facility

Characteristic	Question to Ask
Facility design	Can boarders and clinical cases be housed separately? Is there adequate capacity to house boarders?
Facility operations	Will clinical staff handle boarding animals? Is staffing adequate to cover both clinical and boarding duties? Can staff adequately work with both groups while using some basic infection control practices to reduce the risk of cross-contamination?
Caseload	What percentage of the patient population is at high risk for infection? What percentage of the boarding population may be high risk for pathogen shedding?
Boarding population	Will all animals be accepted or just those deemed low risk for pathogen shedding or development of infection?
Clinical management	What is the risk adversity of the facility and tolerance of the potential for boarders to acquire potential hospital-associated pathogens?

Staff pets

Due to the risk of pathogen exposure if staff are allowed to bring pets to work, pets should not have free access to all areas of the clinic. If staff pets are allowed to accompany their owners, they should be housed in a separate kennel so they do not have direct or indirect contact with other animals, or at a minimum the animals should be restricted to non-clinical areas such as offices. The same criteria listed above for boarding animals and below for clinic pets should be applied to staff pets that are allowed to come to work.

From an infection control standpoint, veterinary clinics should never have a resident "clinic pet".

Clinic pets

Veterinary clinics commonly have resident clinic animals. Although there are no objective data quantifying the risks to patients, people or clinic animals themselves, based on the theoretical risks and lack of a real need for clinic pets, it is recommended that veterinary clinics do not keep such animals, and every attempt should be made to adopt out any existing pets.

While suboptimal from an infection control standpoint, if a clinic has a clinic pet, the following recommendations should be considered:

- Do not allow the clinic pet to have access to any patient treatment areas, patient housing areas, examination rooms, isolation, surgery or the patient waiting area.
- Do not allow the clinic pet to wander freely through the kennel/ward areas where it could cross-contaminate kennels.
- Keep a dedicated food and water bowl, litter box, toys, etc. for the animal.

- The pet must receive regular health checks and have an appropriate vaccination, deworming and external parasite control program.
- Do not allow clinic pets, particularly cats, to have unsupervised outdoor access because of the higher risk of exposure to (and subsequent shedding of) pathogens such as Salmonella and Toxoplasma from hunting birds and rodents.
- Remove the animal from the clinic if any of the following are observed:
  - aggressive behaviour towards people or other animals
  - inappropriate elimination
  - inability to properly restrict access to certain areas
  - development of conditions that pose a risk of pathogen transmission to or from other patients, personnel or clients

Blood donor animals and colonies

Blood donation programs for dogs and cats are increasingly common, and some facilities coordinate their own programs based on colonies of animals (predominantly cats) that live within the clinic. This can be advantageous because of the ready access to screened donors, but there is potential for these animals to both acquire pathogens from the patient population and transmit pathogens to patients. Some basic measures can be used to minimize the risks:

- House donors apart from patients, ideally in an entirely separate room.
- Do not allow donors to roam freely in the facility.
- Use dedicated food and water bowls, litter boxes, toys etc. for donor animals
- Ensure proper quarantine and testing of new animals prior to entry into the colony.
- Schedule socialization activities so that contact with patient animals is avoided.

(See Chapter: Blood Donation for additional recommendations for infection prevention and control regarding blood donation programs.)

Research and teaching animals

It is less common now to have research and teaching animals housed within a teaching hospital, based largely on the evolution of animal care standards, but this practice may still exist in some facilities. In general, this is undesirable because it increases the susceptible animal population in the hospital, establishes a population of long-term residents that could act as sources of infection, and poses a risk to the health of the research colony.

Efforts should be undertaken to remove research animals from hospitals. If the presence of a research or teaching colony is unavoidable, the animals should be physically and procedurally separated from patient animals to minimize direct and indirect exposure. Protocols should be established to require prompt diagnostic testing should any potentially infectious diseases arise in order to reduce the risk to other animals in the facility. Active surveillance for certain pathogens may be necessary depending on the animal source, animal species, pathogen prevalence and other factors. A risk assessment must be performed to determine the optimal isolation, monitoring and testing requirements.

References

Ford RB, et al. 2017 AAHA canine vaccination guidelines. J Am Anim Hosp Assoc. 2017;53:243-51.

Ghosh A, et al. Resident cats in small animal veterinary hospitals carry multi-drug resistant enterococci and are likely involved in cross-contamination of the hospital environment. Front Microbiol 2012;3:62.

Lane HE, et al. 2017. Canine oral papillomavirus outbreak at a dog daycare facility. Can Vet J. 58(7), 747-749.

Weese J, Armstrong J. Outbreak of Clostridium difficile-associated disease in a small animal veterinary teaching hospital. J Vet Intern Med. 2003;17:813-6.

Weese JS, et al. Emergence and containment of Canine Influenza Virus A(H3N2), Ontario, Canada, 2017-2018. Emerg Infect Dis. 2019;25(10):1810-6.

Education

Client education

Client education is the responsibility of the entire practice team. By helping clients understand infectious and zoonotic disease risks and the basic steps they can take to protect themselves and their animals, they can live happier and healthier lives with their pets.

Discussion of zoonotic disease risks should be a routine part of new pet examinations and new client visits. Client education must also occur when the veterinarian has a reasonable suspicion of a potentially infectious disease, and particularly if the disease is zoonotic. Notification of the owner to this effect must be documented in the patient's medical record. This documentation may also be very important legally, should an animal's infection result in human illness.

Items to discuss, information to provide to the client in print form, and/or information to document in the medical record may include:

- what disease is suspected or has been diagnosed
- how the disease is confirmed, if necessary
- how the disease is transmitted
- risks to members of the household
- risks to other in-contact individuals (e.g. those who might visit the household or pet)
- risks to in-contact pets
- basic signs and symptoms in humans (always referring the owner to physician for more specifics)
- clinical signs in animals
- how to prevent disease transmission from the pet to people and to other pets
- how the disease is treated in animals
- public health enforcement issues such as quarantine, submission of tissues to labs, etc. (see Chapter: Reportable Disease)
- circumstances under which the client should seek medical advice, if applicable

Client education is the responsibility of the entire practice team.

Written and online resources for clients for common conditions diagnosed in pets, including some infectious diseases, are increasingly available. Clinics should be prepared to provide clients with or refer clients to resources that have been researched in advance to ensure they contain reliable information that is relevant to the area in which the client lives or travels. This also helps avoid situations in which clients find information on their own which may be inaccurate. See References for examples of websites with free quality client resources.

Veterinary staff education

All veterinary personnel should be familiar with basic zoonotic disease principles and infection prevention and control practices. As infection control is everyone's responsibility, all staff should be educated on clinic protocols relevant to their individual roles. An individual with increased interest in infection prevention and control should take on the role of the clinic infection control practitioner (ICP). This person helps facilitate the infection control program and helps respond to inquiries from other staff members or clients. Any staff member who is unsure of proper infection control procedures should consult the ICP, the clinic infection control manual, or other reference sources (such as these guidelines), to facilitate the use of best practices.

Important principles for the entire practice team to understand (all of which are covered in the respective chapters of these guidelines) include:

- the importance of having an infection control program (the "why")
- hand hygiene
- patient care and handling, including patients that are high-risk for transmitting or acquiring infectious diseases
- appropriate personal protective equipment (including clinic attire)
- cleaning and disinfection of patient waiting areas, exam rooms, and routinely-used equipment
- requirements for sterilization
- sharps safety
- injury prevention

Minors, students, volunteers, and others

Veterinary clinics can provide a rich learning environment for a variety of individuals, including minors with an interest in veterinary medicine, students (e.g. co-op or other), and other volunteers. Training of these individuals should be similar to that of regular staff, but may be limited to very specific duties so as not to overwhelm them. If this is the case, there must be clear limitations regarding in what activities the individual is allowed to engage when unsupervised so that staff and patient safety and infection control protocols are not compromised unintentionally. Use of plain language is especially important for individuals such as these who are likely unfamiliar with medical or other technical jargon. Parental consent is also necessary for anyone who is a minor. Parents must be informed of reasonable risks (infectious disease and other) prior to minors volunteering or working in the clinic, and should provide written consent to this effect.

Documentation of training and consent forms should be kept to avoid potential liability due to adverse events related to exposure to a zoonotic disease, particularly by a volunteer or minor. Clinics should maintain and regularly review their written policies for acceptance of volunteers and others. Consider consulting legal counsel when developing or modifying such policies.

References

Worms & Germs Blog. Promoting safe pet ownership. Available at: http://www.wormsandgermsblog.com.

Center for Food Security and Public Health (CFSPH). Animal disease information. Available at: http://www.cfsph.iastate.edu/DiseaseInfo/index.php.

Pets and Ticks. Evidence-based information on ticks and tick-borne disease of relevance to Canad ian companion animals. Available at: https://www.petsandticks.com/.

The Ohio State University. Disease Prevention at Canine Group Settings. FAQs/FACT documents. Available at: https://vet.osu.edu/preventive-medicine/vpm-research/disease-prevention-canine-group-settings.

Reportable Diseases

Depending on the jurisdiction, certain infectious diseases in animals may be immediately or periodically reportable/notifiable to regulatory bodies at some level either when they are confirmed through laboratory testing, or sometimes when the disease is merely suspected. These diseases vary between countries, and sometimes between jurisdictions within a country, and tend to focus on exotic pathogens and those of significant zoonotic concern (e.g. rabies). Every veterinary clinic should keep an easily-accessible, up-to-date list of reportable/notifiable diseases in the region. The clinic's Infection Control Manual should clearly state the required reporting procedures, including contact numbers for the appropriate animal health and/or public health authorities. In some cases, reporting/notification may be the responsibility of the laboratory that confirms the diagnosis, but in others it may be up to the veterinarian.

Ontario:
Provincial diseases of public health significance (DOPHS) in humans:
https://www.publichealthontario.ca/en/data-and-analysis/infectious-disease/infectious-diseases-monthly

Provincially notifiable hazards in animals:
http://www.omafra.gov.on.ca/english/food/inspection/ahw/aha-regs-hazards.htm

Canada:
Nationally notifiable diseases in humans:
https://diseases.canada.ca/notifiable/diseases-list

Federally reportable diseases in animals:
http://www.inspection.gc.ca/animals/terrestrial-animals/diseases/reportable/eng/1303768471142/1303768544412

US:
Nationally notifiable conditions in humans:
https://wwwn.cdc.gov/nndss/conditions/

Federally notifiable diseases and conditions in animals:
https://www.aphis.usda.gov/aphis/ourfocus/animalhealth/nvap/NVAP-Reference-Guide/Animal-Health-Emergency-Management/Notifiable-Diseases-and-Conditions

See Appendix: Management of Rabies Suspects for case management of rabies suspects.

Clinic Design

Common problems

Clinic design is critical to effectively implement infection control measures. Unfortunately, infection control has not always been considered when designing clinics. Commonly encountered problems include:

- high animal and personnel movement in areas where procedures are performed.
- use of flooring and kennel surfaces that are difficult or impossible to clean and disinfect.
- inadequate (or absent) isolation facilities.
- lack of a separate area to examine or treat animals with potentially infectious diseases.
- lack of sinks in all examination rooms and treatment areas.
- lack of a separate area for diagnostic specimen processing.
- lack of a separate area for staff to eat and store personal items.
- lack of area for reprocessing of equipment and instruments

Practical measures to improve infection control

In established clinics, correcting design deficiencies can be difficult or impossible, and often expensive. However, practical and cost-effective measures can often improve infection control within an existing facility. For example:

- Place alcohol-based hand sanitizers in patient contact areas wherever sink access is inadequate.
- Provide separate refrigerators for:
  - diagnostic specimens.
  - vaccines and medications.
  - food for human consumption.
- Alter personnel and animal movement patterns to reduce direct and indirect contact of relatively healthy patients with sick patients (see isolation section in Chapter: Patient Care and Handling, and Chapter: Hospital Associated Infections and Other Infectious Syndromes).
- Set aside a designated area for staff for eating, drinking and breaks. These activities should not occur in any area where animals or diagnostic specimens may be present.

Exam rooms

Consideration should be given to the standard contents of an exam room and the relative ease with which items and surfaces can be disinfected. A balance needs to be met between infection prevention practices, client perceptions, and animal care.

Hand hygiene stations should be available in the exam room as well as other locations where there is patient contact. If there is no easy access to a sink, then alcohol-based hand sanitizer should be available at a minimum. This enhances ease of use for staff, and it is also positive for clients to see staff practicing good hand hygiene.

It is increasingly popular for veterinary hospitals to employ strategies that reduce pet anxiety in the clinic. These may include using a yoga mat on a stainless steel table, having baskets for cats to lounge in, or using toys to calm patients in the examination room. Proper cleaning and disinfection of non-fabric items, replacement of towels, blankets or mats, and other infection control practices should be considered regarding these amenities to reduce potential pathogen transmission.

Outdoor elimination areas

From an infection control standpoint, elimination areas can be a significant management challenge. It is difficult to balance optimal materials and practicality. Concrete is easiest to clean, but is likely the least ideal from a patient perspective. The substrates that appeal most to dogs (i.e. grass and dirt) are impossible to disinfect. In any case, prompt removal of feces and any other solid waste (e.g. vomitus) is crucial. High-risk patients should be taken to a specific area away from other patients

and common areas for elimination. The area should be disinfected as much as possible to decrease the environmental pathogen load, while being aware of the limitations of any disinfection protocol on the substrate. Gravel and similar substrates that provide excellent drainage can be advantageous, and can be hosed if necessary which has a dilution effect on any contamination that may be present. Putting elimination areas where there is exposure to sunlight for at least part of the day is also beneficial, as the sunlight helps kill pathogens. Grass should also be mowed regularly and close to the ground for the same reason.

New design considerations

When designing new clinics or when undertaking renovation or expansion of existing clinics, use an architect with experience designing veterinary clinics, and emphasize infection control considerations. Consultation or review of preliminary plans by a veterinary infection control expert is also useful. However, critical assessment of plans with an infection control mindset can readily be performed by any veterinarian. Special emphasis should be given to issues such as:

- number and placement of sinks, as a sink should be present in every examination and procedure room.
- overall clinic flow from “clean to dirty”, with isolation areas well removed from other animal housing or procedure areas.
- use of sealed flooring materials that are amenable to frequent cleaning and disinfection.
- selection of furniture materials and other fixtures that are also amenable to cleaning and disinfection.
- separation of animal procedure areas from areas where diagnostic specimens are processed, and from where dental procedures are performed.
- provision of a dedicated “personnel-only” space for breaks, food storage and consumption, and storage of personal items.

Also see Surgical environment and suite design section in Chapter: Surgery.

Vector Control

Some important pathogens can be transmitted by wild rodents (e.g. mice, rats) or insect vectors (e.g. fleas, ticks, mosquitoes, houseflies). A few of these pests can be true carriers of certain pathogens, meaning they can be infected by or incubate particular pathogens, but many of them can also be non-specific mechanical vectors that simply move microbes from one area or surface to another. **Pest management** is an important aspect of effective prevention and control of infectious disease transmission. Pest management practices include:

- examination of animals upon arrival for ectoparasites such as fleas or ticks, and treatment with an adulticidal antiparasitic medication prior to admission if ectoparasites are detected.
- storing food and garbage in metal or thick plastic containers with tight-fitting lids.
- prompt disposal of food waste and other material (e.g. feces) that may attract rodents or insects.
- sealing potential points-of-entry into buildings to prevent invasion by pests. Common methods include the use of caulk, steel wool or mesh wire under doors and around pipes.
- installation and maintenance of window screens to prevent entry of insects into buildings.
- avoiding leaving doors or unscreened windows propped open.
- elimination of potential rodent and insect nesting sites (e.g. clutter, brush or leaf piles).
- removal of standing water (e.g. empty cans, clogged gutters) outside buildings that can otherwise serve as breeding grounds for mosquitoes.

Additional measures may be warranted for the control of specific pests. Consultation with a pest control expert is recommended if a particular infestation is present, or for additional guidance and information.

Ticks

As insect vector ranges expand due to warming seasonal temperatures, so do concerns regarding the spread of the diseases they carry (e.g. Lyme disease caused by *Borrelia burgdorferi,* Rocky Mountain Spotted Fever (RMSF) caused by *Rickettsia rickettsii).* Education is the cornerstone of effective tick control and the prevention of tick-borne infections.

Tick removal and identification plays a role in infection control and in local surveillance. Dogs can be sentinels for tick-borne diseases that can also affect humans, such as Lyme disease (1). Reporting ticks that are found on pets and confirmed cases of tick-borne disease provides valuable data that can also impact public health initiatives.

Checking patients for ticks in the clinic is also important for preventing tick infestations. In many regions the brown dog tick *(Rhipicephalus sanguineus)* is common, and unlike other tick species it can complete its entire life cycle indoors using canine hosts, and thus can infest a building, necessitating environmental control as well.

Tick removal

Ticks require a feeding time of 24-48 hours before they transmit *B. burgdorferi,* and 5-20 hours for *R. rickettsii* (CAPC 2017). Proper removal is important to prevent transmission of bacteria to the pet and infection of the bite site:

- Wear gloves during removal to prevent exposure to the tick's stomach contents in case the tick is crushed, as this can pose a risk of infection to staff even if they are not bitten by the tick. Wash hands after glove removal.
- Forceps, long nosed tweezers or a commercial tick remover are the preferred tools.
- Apply an antiseptic to the skin after removal and wash the removal device.

Proper removal is important to prevent transmission of bacteria to the pet and infection of the bite site.

Other ectoparasites

Some ectoparasites, such as mites and fleas, can be transmitted from patient-to-patient either directly or indirectly. Contact between patients with ectoparasite infestations and others should therefore be avoided. This may involve scheduling appointments for such patients at the end of the day, or asking the owner to bring the pet inside only when the waiting room in empty. After the appointment, all areas with which the patient came into contact should be vacuumed, cleaned and disinfected as per standard protocols. Towels or bedding that were in contact with the patient should be laundered with hot, soapy water. Personnel should wear disposable gloves and a dedicated lab coat or disposable gown when handling an infested patient and should remove these items and perform hand hygiene prior to contact with other patients.

If an infested patient must be kept in the clinic, precautions should be taken to prevent environmental infestation and patient-to-patient transmission. Patients with fleas should be immediately treated with an adulticidal antiparasitic medication on admission, and retreated as necessary. Isolation procedures are likely adequate for patients with mite or flea infestations (see isolation section in Chapter: Patient Care and Handling). All bedding from the isolated animal should be regularly changed and laundered. Cleaning and disinfection should occur as per standard protocols. Patients should be kept in isolation until there is a reasonable degree of certainty that they are no longer infested.

References

Companion Animal Parasite Council (CAPC). Ticks for dogs (updated 12-Apr-2017). Available at https://capcvet.org/guidelines/ticks/. Accessed Dec-2018.

Appendix: Management of Rabies Suspects

Animals with acute neurological disease are commonly encountered in companion animal practice. Although in most developed countries it is rare for these animals to have rabies, rabies must be considered in many situations due to the potential devastating consequences of human exposure to the rabies virus. It is important to err on the side of caution when determining whether to declare an animal a "rabies suspect." A history of rabies vaccination should not be used to rule out the possibility of rabies. Factors to consider if an animal is exhibiting signs consistent with rabies include:

- risk of previous exposure to a rabid animal.
- travel to or importation from areas where rabies is endemic.
- clinical course (rabies typically causes death within 10 days of initial clinical signs in companion animals, and signs are always progressive).
- vaccination history, as it may reduce risk and make rabies a less likely diagnosis. However, no vaccine is 100% effective and other risk factors must be taken into consideration.
- other differential diagnoses that may explain the cause of the neurological signs.

These are recommendations are based on current procedures for rabies response in Ontario, Canada. Veterinary clinics should confirm and follow their own jurisdiction's guidelines.

Local and national requirements regarding potential rabies cases may vary, but every veterinary clinic needs to be aware of proper procedure in its area, which should be prominently displayed for clinic staff, and include current contact information for the appropriate authorities.

If an animal is suspected of having rabies:

1. Notify the owner that rabies is being considered. The owner should be told about the potential for zoonotic transmission, and be asked to make a list of individuals who have been in contact with the animal recently and whether the animal has bitten anyone in the past 10 days. This information should be documented in the medical record.
2. Notify the clinic's Infection Control Practitioner or equivalent.
3. Notify the relevant local animal health and public health authorities. Local and national requirements regarding potential rabies cases may vary, but every veterinary clinic needs to be aware of proper procedure in its area, which should be prominently displayed for clinic staff, and include current contact information for the appropriate authorities.

a. As an example, in Canada rabies is a federally reportable disease, but the response system varies between provinces. In Ontario, in a case of domestic animal exposure to a potentially rabid animal, the attending veterinarian (not the owner) should contact the Ontario Ministry of Agriculture, Food and Rural Affairs (OMAFRA). If there was human exposure, the veterinarian, victim or the victim's physician must contact the local public health unit as soon as possible.

4. Place the animal under strict isolation with a clear warning sign that the animal is not to be handled unless directed by the attending veterinarian. **Entry into isolation and treatment of the patient should be limited to the minimum number of personnel necessary.**

5. Place a “Rabies Suspect” sheet on the cage door. The names of all personnel coming into contact with the animal should be recorded on this sheet.
6. If additional diagnostics or treatments are required, inform all staff that the animal is a rabies suspect. Personnel should not be forced to handle the animal if they are not comfortable doing so. Ideally, only people who have been vaccinated against rabies should be involved in the animal’s care.
7. Avoid procedures likely to result in contact with saliva or cerebral spinal fluid.
8. Anyone handling the animal must wear protective clothing including gloves, gown and face protection. Ensure that any areas of broken skin are securely protected by a bandage or other clothing.
9. Rabid animals can have very unpredictable behavior; employ additional precautions such as the use of catch poles and heavy gloves as needed to reduce the risk of bite injury occurring.
10. Do not euthanize the animal unless it is in extremis, or authorized to do so by the owner and the appropriate authorities.
11. If an individual is exposed through a bite or potential salivary contamination of a wound or mucous membrane:
    a. Immediately and thoroughly wash the wound/area with copious amounts of soap and water. Allow small wounds to bleed to help flush any virus from the tissues.
    b. Apply an antiseptic such as chlorhexidine gluconate or povidone iodine. The rabies virus is very fragile and susceptible to most antiseptics.
    c. Seek medical attention in order to receive rabies post-exposure prophylaxis as soon as possible.
    d. Report all bites to the local public health unit, or other appropriate agency, depending on the jurisdiction.
12. If rabies is ultimately confirmed, public health personnel will determine the need for rabies post-exposure prophylaxis for each individual who had contact with the animal, depending on the circumstances for each.

More information on rabies response in Ontario, Canada can be found on the OMAFRA website (see References below).

References

Brown CM, et al. Compendium of animal rabies prevention and control, 2016. J Am Vet Med Assoc. 2016;248(5):505-17. Available at: http://nasphv.org/Documents/NASPHVRabiesCompendium.pdf. Accessed Dec-2018.

Ontario Ministry of Agriculture, Food and Rural Affairs (OMAFRA). Rabies in Ontario. Available at: http://www.omafra.gov.on.ca/english/food/inspection/ahw/rabies.htm. Accessed Dec-2018.

Surgical Safety Checklist

To be read aloud with the veterinarian and technician present at each stage.

1 Before induction of anesthesia

Patient Check

Patient information confirmed

- Identity _____
- Site and procedure _____
- Owner consent ❑

Required clinical information

- Bloodwork ..... ❑ YES ❑ N/A
- Imaging ..... ❑ YES ❑ N/A
- Other ..... ❑ YES ❑ N/A

Any known allergies ..... ❑ YES ❑ NO

Required medications

- Analgesia ❑
- Antibiotics ..... ❑ YES ❑ N/A
- Other ..... ❑ YES ❑ N/A

IV access and fluids ready ..... ❑ YES ❑ N/A

Difficult airway / aspiration risk

- Yes, and assistance/equipment available ❑
- No ❑

Anticipated blood loss >5%

- Yes, and type-specific blood products available ❑
- No ❑

Patient ASA grade (circle): 1 2 3 4 5

Equipment Check

- Anesthesia equip. safety check completed ❑
- Required monitoring equipment available ❑
  - e.g. pulse ox, ECG, blood pressure, temperature
- Required special instruments/implants available
  ❑ YES ❑ N/A
- Intra-op imaging required/equipment available
  ❑ YES ❑ N/A
- Intra-op sampling required/equipment available
  ❑ YES ❑ N/A
- Suction working ..... ❑ YES ❑ N/A

2 Before skin incision

- Expected length of surgery: _____
- Review critical / non-routine steps expected ❑
- Sterility of surgical packs / kits confirmed ❑
- Swab / sharps count complete ❑
- If significant blood loss is expected,
  blood products available ..... ❑ YES ❑ N/A
- Essential imaging displayed .... ❑ YES ❑ N/A
- Specific patient needs
  - Eye lube ..... ❑ YES ❑ N/A
  - Empty bladder ..... ❑ YES ❑ NO
  - Warming equipment ..... ❑ YES ❑ N/A
  - Antibiotics with 1 hour of incision... ❑ YES ❑ N/A
  - Anticipated timing of additional antibiotic dose
    if required: _____
- "Does anyone have any other questions or concerns before proceeding?" ❑

3 Before patient leaves operating room

- Swab / sharps / instrument count complete ❑
- Specimen labeling complete (read labels aloud)
  ❑ YES ❑ N/A
- Any equipment problems to be addressed
  ❑ YES ❑ NO
- Review key concerns for recovery ❑
  - e.g. oxygen, pain management, glucose / temperature, circulatory support
- Person to contact owner identified ❑

Based on:
https://lafeber.com/vet/wp-content/uploads/Vets-Now-Surgical-Safety-Checklist.pdf
https://www.who.int/patientsafety/safesurgery/checklist/en/

Visit https://oahn.ca/res ources/ipc-best-practices for the complete guidelines.

Clinic Infection Prevention and Control Audit Checklist

Fully Implemented	Partially Implemented	Not Implemented	Not Applicable	AREAS / ITEMS
Hand hygiene:				
				Sink or alcohol-based hand sanitizer stations available in all patient contact areas
				Signage for alcohol-based hand sanitizers with instructions
				Signage for hand washing with instructions
				Minimize jewellery (rings or bracelets)
				No artificial nails / nail extensions / chipped nail polish
				Staff can identify when to use hand hygiene:
				Before and after patient care
				Before aseptic procedures
				Before putting on and after taking off gloves
				After contact with body fluids or mucous membranes
				After contact with contaminated equipment / surfaces
				After personal body functions (i.e. sneezing, coughing, washroom)
				Before eating
Personal protective equipment available:				
				Gloves:
				Household rubber, reusable
				Latex or other, disposable
				Masks:
				Nose and mouth (e.g. surgical) masks
				N95 masks, including fit testing
				Gowns
				Lab coats
				Goggles / eye protection
				Written policies for dress code:
				Available
				Followed
Cleaning / disinfection procedures:				
				Written protocols and procedures for general cleaning and disinfection:
				Available
				Followed
				Appropriate detergents/cleaners available
				Appropriate disinfectant products available for:
				Patient-contact surfaces
				Equipment and instruments

Fully Implemented	Partially Implemented	Not Implemented	Not Applicable	AREAS / ITEMS
Cleaning / disinfection procedures cont'd:				
				Cleaning and disinfection protocol for clippers:
				Available
				Followed
Disinfection / sterilization of medical equipment:				
				Clear cold sterilization protocol in place, specifying proper cleaning before use, product concentration, contact time, interval for changing solution as per manufacture's instructions
				Written protocols for for cleaning semi-critical and critical devices prior to disinfection / sterilization (including disassembly, sorting and soaking, physical removal of organic material, rinsing, drying, physical inspection, wrapping):
				Available
				Followed
				Autoclave:
				Quality control sterility indicators are included in each autoclaved pack
				Biological indicators are periodically used to ensure adequate sterilization and results are logged
				All autoclaved packs are marked with date of autoclaving
Laundry management:				
				Laundry is cleaned on site or by or a commercial service
				Laundry is dried at high temperatures (65-70C)
				Infectious laundry is pre-soaked in bleach solution
				Soiled laundry is transported in a clean manner
				Clean laundry is segregated from soiled laundry
				Hand hygiene station available in laundry area
Waste management:				
				Clear guidelines regarding segregation of waste that is:
				Biohazardous
				Non-biohazardous
Sharps handling:				
				Approved puncture-resistant, labeled containers used
				Containers available in all required areas
				Sharps are disposed immediately after use (not recapped)
				Containers not more than 3/4 filled
Documentation of staff immunization:				
				Rabies
				Tetanus
				Influenza

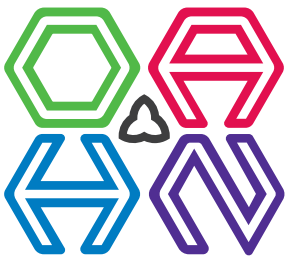
Fully Implemented	Partially Implemented	Not Implemented	Not Applicable	AREAS / ITEMS
Clinic design:				
				Designated isolation area for animals with infectious diseases is available and clearly marked:
				Isolation room vented to outside, or exhaust air HEPA filtered
				Designated diagnostic specimen handling area
				Designated staff “break” area
				Clinic “flow” is clean to dirty
				Separate fridges for food, vaccines and medications, and diagnostic specimens
Vector control:				
				Food debris and clutter eliminated
				Points of entry for rodents and other vermin are sealed
				No standing water outside clinic
				Windows are screened
Examination rooms:				
				Hand washing sinks with soap available in all rooms
				Exam rooms only have essential supplies (no extra storage/stocking)
				Policies followed for cleaning exam rooms:
				Between patients
				At end of day
				Enhanced cleaning/disinfection protocol followed for exam rooms following suspected infectious cases
Isolation area:				
				Isolation area only has essential supplies (no extra storage/stocking)
				Equipment and PPE stay in the isolation area
				Appropriate signage available
				Footbaths or footmats available, if needed
Surgical suite:				
				Surgeon and patient preparation are performed outside the surgical suite
				Personnel traffic in and out of the surgical suite is controlled and minimized
				Surgical suite is only used for sterile procedures (e.g. not dental procedures)
				Surgical safety checklist used for each patient
Staff education and training:				
				Documented annual staff training on:
				Use of personal protective equipment, as applicable
				Cleaning and disinfection protocols, as applicable
				Procedures in the event of potential exposure to zoonotic pathogens, including rabies
				Other infection prevention and control measures, as applicable

Fully Implemented	Partially Implemented	Not Implemented	Not Applicable	AREAS / ITEMS
Staff education and training cont'd:				
				All new staff are provided with infection control training, including:
				How/where to access written IPC protocols (i.e. infection control manual)
				Risk of zoonotic disease exposure, and how to mitigate such
				Documented receipt of training (i.e. signature)
Infection control program:				
				Designated infection control practitioner (ICP) oversees the program
				Surveillance in place (active or passive), as applicable:
				Surgical site infections
				Other hospital-associated infections
				A list of reportable disease readily available at the clinic
				Contact numbers for appropriate veterinary and public health authorities readily available at the clinic
Miscellaneous:				
				No clinic pets
				Policy to not to feed in-patients raw meat diets
				Printed materials or online resources for clients on zoonotic diseases readily available
				Checklist for front office staff when booking appointments to identify potentially infectious animals
				Protocol for advising staff and how to handle potentially infectious patients on admission
				Established policy on admitting animals from shelters or other high-risk sources (e.g. recently imported animals)

Suggested use for audit checklist: The clinic Infection Control Practitioner (ICP) can complete the checklist annually (or more frequently) to document improvements over time, and to identify priority items to be addressed over the next 3, 6 or 12 months. Improvements can be incremental over time. It can also be helpful to have different people complete the checklist and compare the results as a part of quality control.

Notes:

Visit https://oahn.ca/resources/ipc-best-practices for the complete guidelines.



oahn@uoguelph.ca
519-824-4120 x 53364

www.oahn.ca