

zoetis

AIDAP | AUSTRALASIAN INFECTIOUS
DISEASES ADVISORY PANEL

PRACTICAL INFECTION CONTROL GUIDELINES

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The knowledge source for Veterinary professionals

EDITION ONE

ZOETIS LEADING INNOVATION IN ANIMAL HEALTH **FOR OVER 75 YEARS**

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INTRODUCTION

All veterinary hospitals face challenges in preventing the spread of infectious diseases. Hospital-acquired infections result in negative consequences for patients and impact on both the emotions and finances of pet owners. Veterinary hospitals have the potential to both harbour and amplify the spread of infectious agents. This may contribute to the generation of antimicrobial-resistant bacteria and contribute to the spread of these bacteria in the general population.

Poor personal hygiene and suboptimal cleaning and disinfection practices can seriously impact outcomes for patients and lead to resident infectious agents that spread from patient to patient within the hospital environment.

Veterinarians must also consider the very important potential for zoonotic disease and reverse zoonotic disease associated with contact of humans with both healthy and sick animals. Two pertinent and unexpected examples that have emerged over the last 10 years are the occurrence of Q fever (due to *Coxiella burnetii*) after obstetric interventions or caesarean sections and swine brucellosis in pig hunting dogs in northern NSW and Queensland. For these reasons, infection control in veterinary hospitals is of paramount importance. When considered in concert with antimicrobial stewardship, these two measures are absolutely critical to the effective functioning of a modern small animal hospital.

The implementation of infection prevention and control practices in veterinary hospitals is gaining increasing awareness with the profession and the public, especially after the emergence of methicillin-resistant and multidrug-resistant *Staphylococcus* infections in dogs and cats in North America, Europe and more recently in Australia. The purpose of this document is to provide a set of practical guidelines for implementation of infection prevention and control practices in Australian veterinary hospitals.

HOW TO START

Every clinic should develop an **infection control program** and every practice should have a designated **infection control champion/officer**. A written set of guidelines, specific to the requirements of the practice, must be generated. We hope to provide a conceptual framework for this document, and perhaps even a generic template that can be modified to suit the needs of different types of practices. This document will serve as an infection control manual that aids in training new and existing staff and serves as a quality control program. Ongoing surveillance of surgical infection rates; antimicrobial resistance in samples cultured from patients, particularly hospitalised patients, may identify potential breaks in correct procedure.

FOUR GUIDING PRINCIPLES



Identify potential source(s) of organisms.

e.g. skin, wounds, faeces, urine, saliva, blood, aerosols, and discharges.



Decrease transmission.

Consider methods of transmission including hand contact, fomites, droplets, airborne and vector borne infections.



Decrease exposure risks.

Institute awareness programs, ensure appropriate work procedures in place



Maximise patient resilience and immunity

e.g. vaccination/anthelmintic therapy/minimise unnecessary antimicrobial use.

There is a requirement to obtain a microbial diagnosis to the species level and determine the antibiogram of the causative agent if an increase in the prevalence of hospital-acquired infections is discovered.

SECTION 1: HAND HYGIENE

- ⊕ Hand hygiene is the responsibility of all individuals involved in veterinary care.
- ⊕ Veterinarians, veterinary nurses and technicians, receptionists, animal attendants, cleaners and pet owners all have their respective roles to play.
- ⊕ Effective hand hygiene kills, inactivates or removes microorganisms present on the skin while maintaining hand health and skin barrier function (i.e. prevention of chapping and cracking of skin).

Sterilisation of the hands is not the goal of routine hand hygiene, nor is it possible – the objective is to reduce the number of microorganisms on the hands, particularly the number of microorganisms that are part of the transient microbiota of the skin, as these include the majority of potential opportunistic pathogens. These transient microbes may be acquired by contact with a patient, another person, contaminated equipment, or the environment.

There are two methods of removing/killing microorganisms on hands:



washing with soap
and running water



using an alcohol-based
hand sanitiser

AIDAP sees both methods as having a role to play, but our emphasis is on alcohol-based sanitisers.

It is reassuring to the client to see clinic personnel performing hand hygiene, and it increases client awareness of the importance of hand hygiene. Practices may wish to reinforce this by providing alcohol-based hand sanitisers in the waiting room. Clients should also be encouraged to use provided hand sanitisers themselves when they enter the clinic.

ALCOHOL-BASED SANITISERS

Alcohol-based hand sanitisers/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 hand washing with antibacterial soap, can be applied quickly, are less likely to cause skin damage, and can be made available at almost any point-of-care. Their efficacy can be improved further by incorporation of chlorhexidine, which affords residual activity, especially against staphylococcal species. The improved hand hygiene compliance seen with alcohol-based hand sanitisers and their efficacy against most pathogens are important aspects of infection control.

The Hand Hygiene Australia website (<http://www.hha.org.au/>) is a useful resource for further data on importance, research and methods of hand hygiene.

Although alcohol-based hand sanitisers are **not effective against certain pathogens**, including **bacterial spores** (e.g. clostridial spores) and **Cryptosporidium spp.**, routine use of alcohol-based products has not resulted in detectable increases in *Clostridium difficile* spp. infection rates in human hospitals.

If hands are potentially contaminated by one of these organisms, hand washing with soap and running water should be performed if possible.

- + Even though 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 ('the solution to pollution is dilution').

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.

- + As for clostridial pathogens, hand washing with soap and running water is likely more effective, and should be used whenever possible when these pathogens are involved.

Use of products containing **emollients** helps to reduce skin damage which can otherwise occur with frequent use of hand sanitisers.

- + Products containing alcohol and chlorhexidine are also available.
- + Use of **non-alcohol-based waterless hand sanitisers** in healthcare settings is **not recommended**, e.g. sanitisers based on quaternary ammonium compounds (QACs).

PRACTICAL TIP:

If hands are not visibly soiled, clean your hands by rubbing them with an alcohol-based formulation. The ideal alcohol-based hand sanitiser should contain 70–90% v/v alcohol.

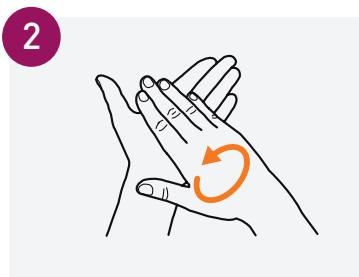
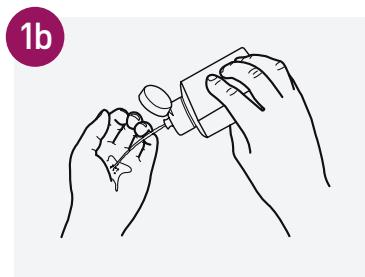
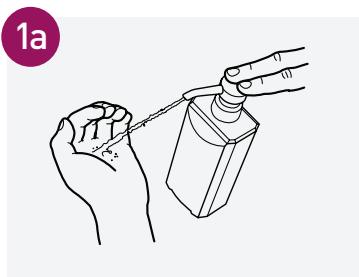
Technique

- 1 Remove all hand and arm jewellery. (As an aside, wearing such jewellery, except possibly a single wedding band, is to be generally discouraged in veterinary hospital, as is the use of nail polish).
- 2 Ensure hands are visibly clean (if soiled, follow hand washing steps).
- 3 Apply **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 20-30 seconds** if sufficient product is used. Hands must be fully dry before touching the patient or patient's environment/equipment for the hand rub to be effective.

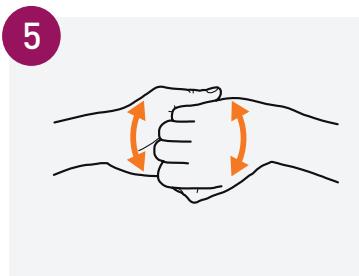
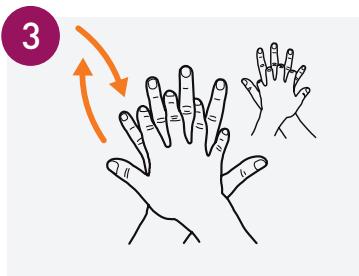
Duration
for the entire
procedure:
20-30 seconds

How to hand rub?

Rub hands for hand hygiene! Wash hands when visibly soiled.



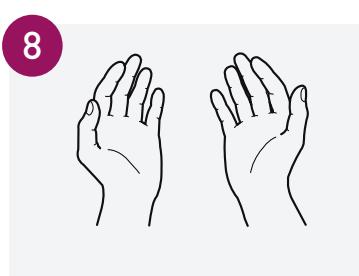
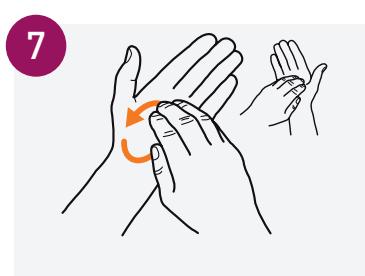
Apply a palmful of the product in a cupped hand, covering all surfaces;



Right palm over left dorsum with interlaced fingers and vice versa;

Palm to palm with fingers interlaced;

Backs of fingers to opposing palms with fingers interlocked;



Rotational rubbing of left thumb clasped in right palm and vice versa;

Rotational rubbing, backwards and forwards with clasped fingers of right hand in left palm and vice versa;

Once dry, your hands are safe.

Adapted from the World Health Organisation (WHO) Guidelines

HAND WASHING

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 or when dealing with a patient with an organism known to be resistant to standard procedures e.g. *Cl. difficile*.

- ✚ If running water is not available, use moistened towelettes to remove all visible dirt and debris, followed by an alcohol-based hand rub.

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

- ✚ Soap should be dispensed via a disposable pump dispenser or automated.
- ✚ Soap containers should not be refilled without being disinfected, since there is a risk of contamination.
- ✚ Antibacterial soaps should be used in critical care areas such as ICU, and in other areas where invasive procedures are performed.

Technique

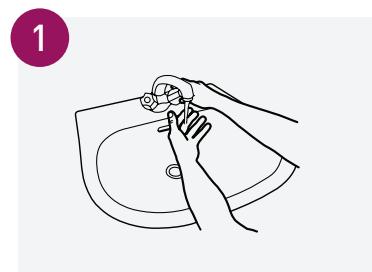
- 1 Remove all hand and arm jewellery.
- 2 Wet hands with **warm** (not hot) water. Hot water is damaging to the skin, and will lead to dryness.
- 3 Apply liquid or foam soap.
- 4 Vigorously lather all surfaces of hands for a minimum of 15 seconds. This is the minimum amount of time required for mechanical 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 the chorus of Waltzing Matilda.
- 5 Using a rubbing motion, thoroughly rinse soap from hands under warm running water. Residual soap can lead to dryness and cracking of skin.
- 6 Dry hands thoroughly by blotting hands gently with a paper towel. Rubbing vigorously with paper towels can damage the skin.
- 7 Turn off taps with paper towel to avoid recontamination of your hands.

PRACTICAL TIP:

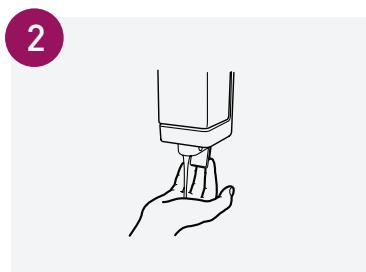
Use "hand-friendly" products e.g. alcohol-based hand sanitizers containing emollients, which can reduce the drying effect of the alcohol.

How to hand wash?

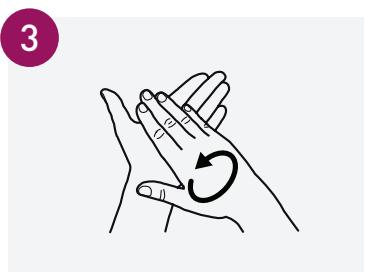
Wash hands when visibly soiled. Otherwise use handrub



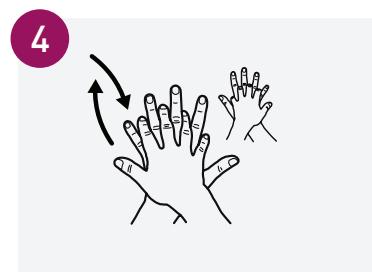
Wet hands with water



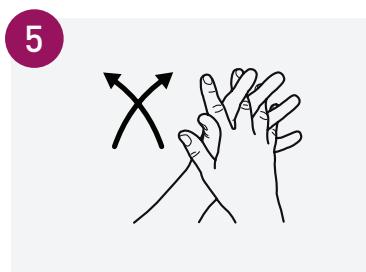
Apply enough soap to cover all hand surfaces;



Rub hands palm to palm;



Right palm over left dorsum with interlaced fingers and vice versa;



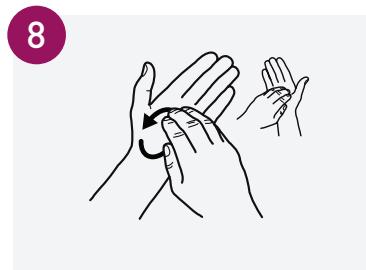
Palm to palm with fingers interlaced;



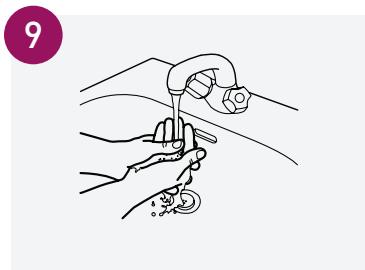
Backs of fingers to opposing palms with fingers interlocked;



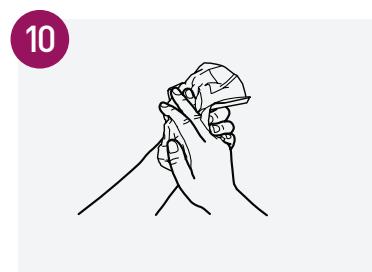
Rotational rubbing of left thumb clasped in right palm and vice versa;



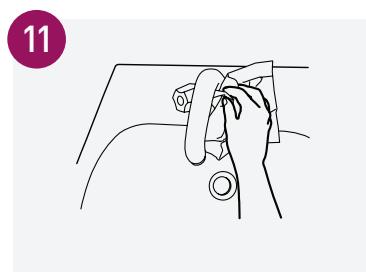
Rotational rubbing, backwards and forwards with clasped fingers of right hand in left palm and vice versa;



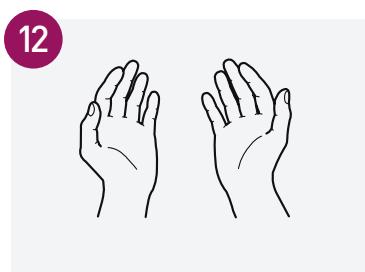
Rinse hands with water;



Dry hands thoroughly with a single use towel;



Use towel to turn off faucet;



Your hands are now safe.

Adapted from the World Health Organisation (WHO) Guidelines

PRACTICAL TIP:

If hands are visibly dirty or visibly soiled with blood or other body fluids, or after using the toilet, wash your hands with soap and water.

FACTORS THAT INFLUENCE THE EFFECTIVENESS OF HAND HYGIENE

- ⊕ **Condition of the skin:** Careful attention to skin and maintaining its integrity is an essential part of the hand hygiene program. Intact skin is easier to clean than skin that is dry, cracked, cut, abraded or otherwise inflamed. Callused hands are more difficult to clean and disinfect. Intact skin is the first line of defense against bacteria. 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. Petroleum-based lotion formulations can weaken latex gloves and increase permeability. Lotions that contain petroleum or other oil-based emollients should only be used at the end of the work day. If lotions are used during the work day, select a water-based product.
- ⊕ **Finger nails:** Natural nails more than 3-4 mm long are difficult to clean, can pierce gloves and harbor more microorganisms than short nails. Artificial nails or nail enhancements (including nail polish) should not be worn by anyone involved directly in veterinary patient care, as they have been implicated in the transfer of microorganisms in human medicine. Loose tags of skin or cuticle around the nails or nail bed also provide a potential nidus of organism carriage.
- ⊕ **Jewellery:** Jewellery is very hard to clean, and physically protects bacteria and viruses from the antiseptic action of alcohol-based hand sanitisers and the mechanical cleaning action of soap and running water. Rings and bracelets should not be worn during patient contact. Rings, in particular, increase the number of microorganisms present on hands and increase the risk of tears in gloves.

SECTION 2: PERSONAL PROTECTIVE EQUIPMENT

The use of personal protective equipment (PPE) is required to prevent contamination of personal clothing, reduce exposure of skin and mucous membranes of veterinary staff and reduce transmission of pathogens between patients by veterinary staff. PPE use does not eliminate the need for appropriate environmental controls such as adequate ventilation, isolation rooms etc., nor does it negate the need for hand washing/hand rubs following the procedure.

In veterinary practice, normal clothing worn to and from work should always be covered or replaced by protective wear such as scrubs or a lab coat while working in the hospital or the field.

The following pertains to specific articles of protective clothing:

LABORATORY COATS/SCRUBS

These are commonly used in veterinary clinics and are a basic form of covering for the upper body preventing the soiling of street clothes. These are generally not fluid-resistant and this should be kept in mind where soaking or splashing of infectious material may occur.

- + If soaking or splashing occurs, the coats should be removed and replaced.
- + They should never be worn outside the hospital/clinic.
- + They should be laundered daily.

NON-STERILE GOWNS

Gowns are used for handling animals strongly suspected of having contagious diseases such as in isolation wards. Permeable gowns may be re-used but are typically disposed of after use. Disposable waterproof gowns may be used if splashing with body fluids is anticipated. If re-usable gowns are used, then appropriate disinfection and washing is required after use.

Gloves are used in all situations where gowns are worn. Disposable gowns should be removed before gloves and placed into a waste disposal receptacle suitable for disinfection/decontamination procedures. In situations where underclothing is contaminated, it should be removed and the skin washed and treated with an appropriate disinfectant. Contaminated clothing should be bagged in a non-porous container then treated with an appropriate disinfectant agent (see <http://www.ava.com.au/biosecurity-guidelines>).

PRACTICAL TIP:

There is a great method called the BEAK method, which minimises splashes from glove removal and minimises user contact with fluids. It's readily available via YouTube and recommended by the CDC.

https://youtu.be/B0Ab_cy3HxM

GLOVES

Gloves should be worn in all cases where contact with blood, body fluids, secretions or excretions that are suspected to contain infectious agents occur. They should also be worn during cleaning of cages, examination tables or handling clothing which has contacted suspected infectious material.

- ✚ Remove immediately after use, avoiding contact of outer surface with skin or clothing.
- ✚ Gloves should never handle shared surface objects (phones, pens, paper).
- ✚ Hand hygiene (washing) should always be performed after glove removal.
- ✚ Disposable gloves are only ever used once.
- ✚ Gloves should be changed prior to moving to a 'clean' area or the next patient.
- ✚ Latex gloves are used in most cases, but if incompatible with cleaning or disinfecting agents, then nitrile gloves should be used, or rubber gloves may be used and disinfected. Rubber gloves are not suited to handling infectious materials such as parvovirus-containing faeces.

FACE PROTECTION

Prevents exposure of the mouth, nose and eyes and is critical in situations where droplet, nebulisation or splashing is a potential problem. For example:

- ✚ Dental scaling and prophylaxis.
- ✚ Necropsy procedures, particularly use of saws.
- ✚ Wound lavage.
- ✚ Surgery where infected body fluids may be splashed.
- ✚ This is more important for certain pathogens with inherently greater virulence for normal human hosts e.g. *Brucella suis*, *Coxiella burnetii*.

RESPIRATORY PROTECTION

- ✚ Required for any infectious agent or material that may be transferred by aerosol.
- ✚ Examples of specific masks included the N95 masks.
- ✚ N95 respirators have two advantages over simple cloth or surgical masks; they are >95% efficient at filtering 0.3 µm particles (smaller than the 5 µm size of large droplets created during talking, coughing and sneezing, which usually transmit influenza, for example, in humans) and are fit-tested to ensure that infectious droplets and particles do not leak around the mask
- ✚ For some situations, for example a necropsy on animal(s) suspected to have died from rabies, Australian bat lyssavirus, Hendra virus, or herpes virus B (from primates), then the use of Class 3 Biosafety cabinets are required. Therefore, these procedures should only be conducted in specialist facilities.



N95 Mask/respirators

FOOTWEAR

Footwear should always be of a closed variety and impermeable to fluids. In some situations, to prevent transfer of infectious agents from one location to another, such footwear may be in the form of shoes (or shoe covers) that do not leave the isolation room (and are immediately disposed of). Everyday footwear may be left at the entrance to the isolation room on the “contaminated” side.



FOOTBATHS AND FOOT MATS

These are often used to reduce microbiological content on footwear. Footbaths are shallow containers containing a disinfectant solution. Foot mats are made of spongy material that is saturated with disinfectant solution. Footbaths are often associated with spillage and can create a slippery surface. These should be considered in areas where staff may traffic infectious agents from an isolated/contaminated environment to the general ‘clean’ areas of the hospital.

- ✚ The solution should be changed daily as persistent organic matter within disinfectants reduces their efficacy and some disinfectants are required to be freshly diluted to remain active against organisms.
- ✚ The disinfectant solutions used should be appropriate for the infectious agents likely to be encountered.
- ✚ Care should be taken due to workplace health and safety (WHS) implications associated with slippery floors and wet footwear.

TABLE 1. SELECTION OF APPROPRIATE PROTECTIVE EQUIPMENT RELATIVE TO RISK

Risk Level	Description	Suggested PPE and disinfection
 Low Risk	Routine animal examinations	Hand washing after contact, possible glove use, scrub tops.
 Mild Risk	Handling animals with potential exposure to infectious material e.g. Methicillin-resistant <i>Staphylococcus Pseudintermedius</i> (MRSP), feline herpesvirus type 1	Appropriate PPE and decontamination. This will vary depending on the situation but may include the use of gowns and gloves. Decontamination should involve removal of infectious material by disposal of gloves and gowns. Appropriate hand washing after contact with each animal.
 High Risk	Potential exposure to infectious material that is extremely contagious (CPV) or dangerous. Includes zoonotic diseases e.g. Q fever	High level PPE and decontamination should be employed.
 Very High Risk	High level exotic zoonotic diseases e.g. rabies	Contact a government veterinarian for advice on how to proceed. National Emergency Animal Disease Hotline is 1800 675 888.

Adapted from Ryan and Jacobsen (2009) and is used with the permission of the New South Wales Department of Primary Industries.

SECTION 3: ENVIRONMENTAL HYGIENE

Environmental hygiene is a term that encompasses measures to reduce the overall burden of infectious disease agents in the clinic and hospital. In particular, this involves specific cleaning and disinfection measures.

COMBINING CLEANING AND DISINFECTION

Cleaning should always precede disinfection to prevent organic matter accumulating that may hinder successful use of a disinfectant. Cleaning involves the removal of visible organic matter using soap or detergent. Disinfection involves the use of a chemical or other procedure that kills or inactivates the remaining microbes.

Methods of cleaning and disinfection vary depending on the surface or equipment to be used. For example, cage or flooring requires different methods to specific equipment such as endoscopes and clippers. Staff responsible for cleaning and disinfection tasks should be properly trained and familiar with chemicals and be aware of the importance of this task. Material Safety Data Sheets should be accessible for all chemicals used in the clinic and standard operating procedures provided to ensure the practice meets workplace health and safety regulations and ensures correct infection control practices.

CLEANING

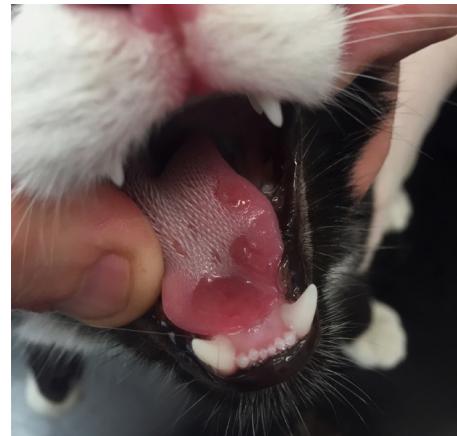
Organic matter such as faeces, blood, urine, dirt, tissue, cat litter or food should be removed from surfaces.

- + Loose dust/dirt may be removed with a cyclonic vacuum cleaner containing a HEPA filter.
- + Sweep with broom prior to mopping.
- + Mops should be cleaned, disinfected and dried at least daily and changed weekly unless there is a significant contamination event when they should be discarded immediately in contaminated waste.
- + Use appropriate protective clothing where splashing/splatter is possible.
- + Use a face mask where aerosol generation and exposure is likely.
- + Adherent material may require scrubbing with detergent or soap.
- + Pressure washers should be avoided due to aerosol generation.
- + Hoses producing less than 120 psi are not routinely associated with aerosol generation.
- + The hand-held portion of a household 'steam mop' can play a useful role in the disinfection of certain surfaces and objects. Commercial steam generators are generally cost-prohibitive.

DISINFECTION

Follow cleaning, disinfection procedures should then be undertaken. The area should be well ventilated and gloves should be worn when handling the disinfectant. Disposable gloves may be used but latex gloves typically deteriorate rapidly and nitrile gloves (purple) are more robust and longer lasting. Rubber gloves that are reusable are preferred and should be disinfected at the end of the task and hung to allow drying. Protective eye wear should be considered where there is a risk of splashing.

- ⊕ Always apply disinfectant at correct dilution, appropriate to both the disinfectant agent and target agent of disease e.g. the final concentration of some disinfectants need to be higher for canine parvovirus than for other agents of disease.
- ⊕ Always apply for recommended contact time, appropriate to the disinfectant and target agent of disease.
- ⊕ Rinsing is required where patients or personnel may come into contact with residual disinfectant agents. This is especially important for cleaning cages which will house cats, as their tendency to groom can lead to tongue ulceration after application of quaternary ammonium compounds.
- ⊕ Allow surfaces to dry completely prior to patient exposure.



Cat with ulcerated tongue and nasal philtrum.

This cat was diagnosed as part of a recent outbreak of virulent calicivirus in Sydney.
(Image courtesy of Dr. David Hughes,
Concord Veterinary Hospital)

ISOLATION WARDS

An isolation ward (ISO-W) to house and treat animals with contagious infections is essential for all veterinary hospitals. An ISO-W should ensure physical separation of patients with contagious diseases from other hospitalised patients. The ISO-W should be in a low-traffic area that is accessible without having to walk through treatment areas or general hospital wards.

- ⊕ A separate entrance to the main hospital to allow animals suspected to have a contagious disease during consultation to be admitted directly to the ISO-W for any treatment is desirable.
- ⊕ Wherever possible, the ISO-W should be ventilated separately to the rest of the hospital, with extraction fans directing airflow directly outdoors.

All staff should be inducted and trained on the standard operating procedures (SOP) of the ISO-W. The SOP document should be readily available. The ISO-W should be clearly identifiable to all staff using signage on display near the entrance (e.g. door). Signs should include reference to use of appropriate PPE and the practice's biosecurity protocol.

- ⊕ Ideally in large vet hospitals a log-book to record name and time of all staff entering and leaving the ward should be available in an easily accessible location outside the ISO-W.
- ⊕ Only staff essential to patient management (and not including owners of hospitalised patients) should enter the ISO-W.

The ISO-W should not be used as a storage area for non-essential equipment. It should be easy to clean and disinfect and only contain equipment required to manage patients. Equipment in the ISO-W should be cleaned and disinfected each time it is used (e.g. stethoscope, thermometer) and should not be removed from the ISO-W except to be disposed of via contaminated waste disposal services. Supplies for use in future patients, e.g. gauze swabs and bandaging material should not be stored in the ISO-W unless stored in small amounts in enclosed cupboards or containers.

PRACTICAL TIP:

Staff should be notified immediately if the waiting room and consulting room become contaminated so they can be cleaned and disinfected.

Recommended equipment in the ISO-W

- + Sharps container.
- + Waste container lined with a plastic bag then double-bagged prior to disposal.
- + Contaminated waste container lined with a contaminated waste bag.
- + Dedicated cleaning equipment.
- + Laundry container lined with a contaminated waste bag.
- + Stethoscope and thermometer.
- + Scissors.
- + IV-fluid pump.
- + Clippers.

Footbaths or foot mats (see PPE) should be used before entry to and after leaving the ISO-W. Barrier nursing is mandatory for all patients in the ISO-W and appropriate protective equipment should be worn (see PPE section). Double-gloving (wearing an inner and outer pair of nitrile gloves) is recommended (see video resource www.ava.com.au/suit-up). Note that gowns should be used once then disposed of or laundered at a minimum temperature of 40°. When a patient is discharged from the ISO-W, all items remaining should be disposed of or disinfected. All waste should be treated as potentially infectious. The room should be cleaned twice.

Staff laundering bedding, gowns or other items from the ISO-W should follow standard hand hygiene protocols while handling these items and wear appropriate PPE. Laundry items should be washed separately to other hospital laundry. For disinfection, the washing cycle should include detergent and bleach. Laundry should be dried in a hot air drier.

MANAGING PATIENTS IN THE ISOLATION WARD

Patients in the ISO-W should be treated after immunosuppressed patients and the use of PPE should be considered as both protective for the ISO-W and the immunosuppressed patient.

Whenever possible, patients should not leave the ISO-W unless they are being discharged. Dogs should not be walked for exercise during the period of isolation and should only leave the ISO-W for essential diagnostic procedures that cannot be performed within the ISO-W (e.g. radiology).

Diagnostic specimens (e.g. faeces, blood) collected into leak-proof containers (e.g. plastic vials) should be placed in a plastic specimen bag that is disinfected before leaving the ISO-W and then double-bagged for dispatch to the laboratory.

In general, patients should urinate and defaecate in the ISO-W. Where this is problematic, dogs should be taken outside via the most direct route and ideally not via high-traffic areas or past other hospitalised patients.

The outside area should be separate to elimination areas for other patients. Faeces should be disposed of with other contaminated waste from the ISO-W and any non-porous surfaces (e.g. cement) the patient has been in contact with require thorough disinfection afterwards.

PRACTICAL TIP:

Good communication between staff is essential. In large hospitals, an email or text message could be used as a quick way to notify all staff if there is suspicion of a notifiable infectious disease. (For further information on notifiable diseases in Australia see <http://www.agriculture.gov.au/pests-diseases-weeds/animal/notifiable>.

DISINFECTANT SELECTION

The selection of disinfectants should take into account the product's spectrum of activity, the capacity for inactivation by organic matter and the potential pathogens that are likely to contaminate the hospital environment. Some disinfectants are not compatible with soaps and detergents, or have toxicity issues for personnel and patients. Some are potentially corrosive or have negative effects on the environment if disposal is carried out. Table 2 characterises the properties of various different disinfectant classes. Some disinfectant compounds are formulated to contain several compound classes. An example is F-10 that contains surfactants, polyhexanide biguanidine and benzylammonium chloride (QAC).



**An example:
F10 veterinary disinfectant**

TABLE 2. CHARACTERISTICS OF SELECTED DISINFECTANTS

DISINFECTANT CATEGORY	ACTIVITY IN ORGANIC MATTER	ADVANTAGES	DISADVANTAGES	PRECAUTIONS	COMMENTS
Alcohols Ethanol Isopropyl alcohol	Rapidly inactive	Fast action No residue Relatively non toxic	Rapid evaporation	Flammable	Primarily used as a topical antiseptic and not as an environmental disinfectant
Aldehydes Formaldehydes Glutaraldehyde	Good	Broad spectrum Non corrosive	High Toxicity	Carcinogenic Irritant Ventilation required	Used as aqueous solution most commonly Cidex™ used for endoscopy disinfection is an example
Alkalies Ammonia			Unpleasant odour	Incompatible with bleach solutions	Not suggested for general use
Biguanides (including polyhexanide chlorhexidine)	Rapid inactivation Biguanides can only function at pH range (5-7) Inactivated by soaps and detergents	Non toxic	Not compatible with anionic detergents		Primary uses as antiseptic not an environmental solution
Halogens Hypochlorite (bleach)	Rapid inactivation	Broad spectrum Sporicidal, virucidal, bactericidal Suitable for food preparing surfaces	Inactivated by cationic soaps and detergents Frequent application needed Fresh solutions required as diluted disinfectant unstable	Corrosive irritant May produce chlorine gas if mixed with other chemicals such as ammonia	Used to disinfect and clean environmental surfaces
Oxidising agents e.g. potassium peroxyomonosulfate	Good	Broad spectrum Environmentally friendly	Breakdown with time	Corrosive	Excellent choice for environmental disinfection
Phenols	Good	Broad spectrum	Toxic especially to cats Horrible odour Incompatible with non-cationic and non-ionic detergents	Irritant	Some residual activity after drying
Quaternary ammonium compounds (QACs)	Moderate	Stable storage Minimal skin irritation Low toxicity Can be used on food preparation surfaces Effective at high temperatures and pH	Incompatible with anionic detergents		Commonly used primary environmental disinfectant

Prepare and use all disinfectants as directed on the product packaging. Products may not be effective if prepared or used incorrectly. Do not combine disinfectant products or combine disinfectants with soap and detergents in a single solution, unless using solutions commercially prepared for this purpose.

TABLE 3. COMMONLY USED DISINFECTANTS

DISINFECTANT	5.25% HOUSEHOLD BLEACH (SODIUM HYPOCHLORITE)	QUATERNARY AMMONIUM COMPOUNDS (QAC) /BIGUANIDE COMBINATIONS	ACCELERATED HYDROGEN PEROXIDE	POTASSIUM PEROXYMONO-SULFATE
Trade names	White King, Clorox	F10, Trigene II		Trifectant, Virkon S
Effective against parvovirus?	Yes, dilute 1:30	Yes, check recommended dilution	Yes	Yes
Effective against dermatophytes?	Yes, dilute 1:10	Yes, check recommended dilution	Not known	No
Effective against calicivirus?	Yes, dilute 1:30	QAC have variable efficacy against calicivirus depending on what they are combined with - they are not first choice. F10 does not protect against calicivirus at any dilution	Yes	Yes
Inactivation by organic matter?	Yes	Moderate inactivation	No	Mild inactivation
Stability when prepared?	24 hours (if kept dark)	Supplied ready to use	Supplied ready to use	7 days
Contact time required	10 minutes	2-30 minutes depending on indication	5-10 minutes	
Detergent required?	Yes prior	No but rinsing required	Yes	Some intrinsic activity for mild organic matter
Cost	Low cost	Moderate	Expensive	Moderate
Notes	Corrosive to metals	F10: General disinfection dilute 1:500, 1:100 for high risk. Trigene II: General disinfection dilute 1:100, Used as supplied dilute 1:50 high risk or soiled areas. (Read labels for more details)		Made up from powder in some cases. Can be corrosive

Disclaimer: The use of trade names does not in any way signify endorsement of a particular product

TABLE 4. ANTIMICROBIAL SPECTRUM OF SELECTED DISINFECTANTS

	AGENT	ALCOHOLS	ALDEHYDES	ALKALIS: AMMONIA	BIGUANIDES: CHLORHEXIDINE	HALOGENS: HYPOCHLORITE (BLEACH)	OXIDISING AGENTS	PHENOLS	QUATERNARY AMMONIUM COMPOUNDS
Susceptibility of microorganisms to chemical disinfectants.	Mycoplasmas	++	++	++	++	++	++	++	+
	Gram-positive bacteria	++	++	+	++	++	++	++	++
	Gram-negative bacteria	++	++	+	+	++	++	++	+
	Pseudomonas	++	++	+	±	++	++	++	+
	Enveloped Viruses	+	++	+	++	++	++	+	+
	Chlamydiae	+	+	+	±	+	+	-	-
	Non-enveloped Viruses	-	+	±	+	++	+	+	-
	Fungal spores	+	+	+	±	+	+	+	+
	Acid-fast bacteria	+	++	+	-	+	+	+	-
	Bacterial Spores	-	+	±	-	++	+	-	-
Most resistant	Coccidia	-	-	-	-	-	-	+	-

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

Source: Canadian Committee on Antibiotic Resistance 2008, Infection prevention and control best practices for small animal veterinary clinics, Canadian Committee on Antibiotic Resistance, Guelph, Ontario.

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:** *Chlamydia 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*

*In general, phenols are not effective against non-enveloped viruses, but they have been found to be effective against rotaviruses. They have been recommended for use on horse farms to help control equine rotaviral disease in foals. However, efficacy against small animal parvoviruses has not been demonstrated.

MISCELLANEOUS ITEMS

- ✚ Food and water bowls should be washed and cleaned prior to disinfection. Sharing of unused food bowls should never occur.
- ✚ Thermometers should be cleaned with disposable paper toweling prior to disinfection with alcohol-based disinfectants and allowed to dry.
- ✚ Cat litter boxes should be cleaned and disinfected (soaked or thoroughly wiped down) daily, or after defaecation.
- ✚ Cat litter boxes should ideally not be handled by pregnant women. This may be safe, however, if the litter is cleaned daily prior to sporulation of *Toxoplasma* oocysts.
- ✚ Stethoscopes should be wiped between patients using an alcohol based disinfectant.
- ✚ Clean clippers in between use and have separate clippers for dirty and clean procedures.

SECTION 4: GENERAL PROCEDURES

INTRODUCTION

In veterinary practice it is important to consider that every patient that enters an examination room carries the risk of harbouring a communicable agent of disease. Careful attention to disinfection of the examination room, equipment and an understanding of the transmission of infectious agents can limit the risk of spread of disease. This section describes procedures for disinfection of examination rooms and equipment.

CLEANING OF EXAMINATION ROOMS

Ideally, examination rooms should have no porous or irregular surfaces that support niche environments for organisms. Typically most veterinary hospitals will have tiled or linoleum covered floors. Tables may be made of stainless steel or may have Formica coverings. Consulting tables should be cleaned and disinfected between each patient. A suitable agent is F-10 and disposable paper towels. If tablemats are used to obtain stable footing for patients these should be replaced daily and disinfected after each patient. For feline patients, a fresh towel on the table for each patient is a good measure. Faecal, urinary or other body fluid (blood or wound discharge) contamination should prompt immediate replacement followed by cleaning and disinfection.

Thermometers should be cleaned immediately after use by wiping with a disposable paper towel to remove organic matter and then wiped with disinfectant soaked paper towel. Storage should be in a dry covered case.

Floors should be swept or vacuumed (cyclonic unit with HEPA filter to stop circulation and aerosolisation of fine infectious particulate material) then mopped wet with detergent/disinfectant as appropriate between each consulting period. It is important to remove all waste and dirt prior to application of disinfection agents. The mop and bucket should be drained, rinsed with water until clean after use and stored dry (preferably in the sun for its UV content) due to the antimicrobial effects of UV light.

Surfaces that are frequently handled including computer keyboards, smart phones, light switches and door handles should be wiped clean several times daily using paper towel soaked in disinfectant or a soft disposable cloth. These can be dried with a clean paper towel afterwards. Sinks should be cleaned daily using a cleanser and disposable cloth or towel and then disinfectant applied to the sinkhole.

Spray bottles and soap dispensers need to be disinfected between each refill to prevent recontamination. Wiping the surface of the dispensers and bottles with paper towels and disinfectant is recommended on a daily basis.

PRACTICAL TIP:

If using vertical holders containing disinfectant then the disinfectant must be changed on a regular basis or organisms such as *Pseudomonas* may eventually grow in the solution.



Clean the thermometer immediately after use

(Photo courtesy of Dr. Bronwyn Walker)

CLEANING OF STETHOSCOPES AND SMART DEVICES

The surface of the stethoscope should be wiped with a paper towel soaked in disinfectant following each use. Consider getting a waterproof case for the clinic smart devices so they can be easily cleaned and disinfected.

PRACTICAL TIP:

Nappy wipes could be an alternative to paper towels soaked in disinfectant to improve staff compliance.



CLEANING OF OTOSCOPES

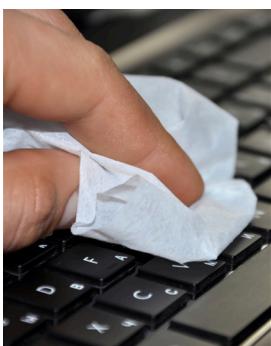
Most veterinary otoscopes come with reusable specula that are lightweight yet durable. These may be autoclaved, boiled, or sterilised in a suitable solution to achieve sterility. In most instances, cleaning, removal of wax and other debris is followed by the use of a topical disinfection agent between patients. If the ear contains large amounts of debris or infectious material then cleaning the otoscope should be performed followed by immediate topical disinfection and drying prior to examining the other ear. Suitable cleaning agents include chlorhexidine soaps such as those used for hand cleansing. This may be followed by immersion in a disinfection agent such as alcoholic chlorhexidine and drying. Otoscope specula should not be kept soaking in disinfectants as important agents of disease in otitis externa e.g. *Pseudomonas aeruginosa* will persist in disinfectant solutions.

PRACTICAL TIP:

Ideally there should be sufficient numbers of specula to allow autoclaving before reuse. If it is not possible then cleaning, removal of wax and other debris should be performed followed by the use of a topical disinfection agent between each patient.

CLEANING OF VIDEO-OTOSCOPY UNITS

Some models can be autoclaved and this is the ideal sterilisation method. If it is not possible to autoclave the unit then it should be cleaned to a high level of disinfection in accordance with the manufacturer's recommendations, because of the risk of serious contamination and transfer of resistant bacteria such as MRSP and *Ps. aeruginosa*. The instrument should be soaked until it is cleaned to prevent any organic material drying on the surface. It should be thoroughly cleaned with a soapy solution, including the working channel. The instrument is then soaked in a cold sterilising solution according to the individual manufacturer's recommendations (e.g. glutaraldehyde or ortho-phthalaldehyde OPA). Once soaked, it should be thoroughly rinsed with plain water and dried prior to subsequent use.



CLEANING OF DIAGNOSTIC EQUIPMENT (ULTRASOUND MACHINES, RADIOGRAPHY MACHINES)

Ultrasound probes should be cleansed of coupling gel after each procedure and wiped clean. Use cleaning methods as recommended by the manufacturer. Keyboards/trackpads should also be cleaned on a regular basis. Following examination of the animal the consultation and room should be cleaned between patients.

ANAESTHETIC EQUIPMENT DISINFECTION

As most endotracheal (ET) tubes are reused, they must be subject to high-level disinfection or sterilisation. Ethylene oxide and glutaraldehyde are effective, but may not be available or practical. Thus, at a minimum, the tubes must be washed inside and out in hot soapy water as soon as they are removed from the patient (to prevent any debris from drying on surface) and then soaked in a quaternary ammonium fresh solution, rinsed and dried before use. For animals with a potentially infectious respiratory disease, placement of fresh filter device is recommended to prevent aerosolisation and contamination of the anaesthetic machine.

CLEANING OF ENDOSCOPES

Veterinarians use endoscopes to diagnose and treat numerous disorders. In human medical practice, more healthcare-associated outbreaks have been linked to contaminated endoscopes than to any other medical device. Heat-sensitive endoscopes (e.g. gastrointestinal endoscopes, bronchoscopes, cystoscopes) require cleaning and disinfection after each use. Appropriate use of cleaning and disinfection can be expected to destroy all vegetative microorganisms.

Flexible endoscopes, particularly gastrointestinal endoscopes, may acquire high levels of microbial contamination. Cleaning reduces the level of microbial contamination by around 4 to 6 logs. Similarly, disinfection is only effective when the device is first properly cleaned prior to disinfection.

Suitable formulations for disinfection include: >2.4% glutaraldehyde, 0.55% ortho-phthalaldehyde (OPA), 0.95% glutaraldehyde with 1.64% phenol/phenate, 7.35% hydrogen peroxide with 0.23% peracetic acid, 1.0% hydrogen peroxide with 0.08% peracetic acid, and 7.5% hydrogen peroxide.

- + These products have excellent antimicrobial activity.
- + Users should check with device manufacturers for information about any disinfectant's compatibility with their endoscope.

Ethylene oxide sterilisation of flexible endoscopes is infrequent because it requires a lengthy processing and aeration time (over 12 hours) and would pose a potential hazard to staff and patients. As such, this method of sterilization is not used to any great extent in veterinary practice, unless there is a relationship with a near-by human facility that offers this service. In human medicine, ortho-phthalaldehyde has begun to replace glutaraldehyde in many health-care facilities because it has several potential advantages over glutaraldehyde: is not known to irritate the eyes and nasal passages, does not require activation or exposure monitoring, and it has a 12-minute high-level disinfection claim in the United States. Cidex-OPA is an available example that is suitable for veterinary endoscopes.

Regular disinfectants should not be used for reprocessing endoscopes. This includes iodophors, chlorine solutions, alcohols, quaternary ammonium compounds and phenolics. These solutions should be strongly discouraged because of lack of proven efficacy against all microorganisms or have materials incompatibility (and might thus damage the scope).

Flexible endoscopes are particularly difficult to disinfect and easy to damage because of their intricate design and delicate materials. Meticulous cleaning must precede any sterilisation or high-level disinfection of these instruments. Care with pressure testing for leaks prior to cleaning is important.

Establishment of correct connectors between the devices is critical to ensure complete flow of disinfectants and rinse water through the scope. Suction and insufflation valves should be cleaned and endoscopic accessories should be used according to the manufacturer's instructions. The biopsy and air channels should be cleaned using the specifically-designed accessories provided with the scope to remove all foreign matter before the disinfection process begins.

Practices should recognise that all nursing personnel require training procedures for correct use of the cleaning equipment. To ensure reprocessing personnel are properly trained, each person who reprocesses endoscopic instruments should receive initial and annual competency testing.

Endoscope disinfection with a liquid chemical agent involves five steps after leak testing:

- 1 **Clean:** mechanically clean internal and external surfaces, including brushing internal channels and flushing each internal channel with water and a detergent or enzymatic cleaner (leak testing is recommended for endoscopes before immersion). A commonly used cleansing agent is Cidezyme® that contains KOH and surfactant agents.
- 2 **Disinfect:** immerse endoscope in high-level disinfectant and perfuse to eliminate air pockets and ensure contact of the agent with the internal channels. Using the suction specifically designed attachments, the biopsy channel and air/water channel are perfused for the time recommended for the specific disinfection agent being used. Cidex or Cidex OPA are available options.
- 3 **Rinse:** rinse the endoscope and all channels with sterile water, filtered water or clean tap water. For scopes requiring complete sterility (e.g. bronchoscopes used for BAL), sterile water may be required.
- 4 **Dry:** rinse the insertion tube and inner channels with alcohol, and then provide a final flush with air.
- 5 **Store:** store the endoscope in a way that prevents recontamination and promotes drying (e.g. hung vertically). In addition to the endoscope reprocessing steps, a protocol should be developed that ensures the user knows whether an endoscope has been appropriately cleaned and disinfected or has not been reprocessed. For example if endoscopes are stored/rehung without labeling, confusion can result about whether the endoscope has been processed.

If endoscopes are transported off site for use they should not be returned in their original storage case but transported in a suitable container that can be also cleaned and sterilized. Cidex tray systems are an example of a suitable container that may be used for the manual cleaning and disinfection procedure and also transport from a remote site after use. The practice should designate an infection-control officer to ensure that there is compliance with correct procedures where endoscopes are reprocessed.

SURGERY

All surgical procedures are accompanied by an inherent risk of surgical site infection (SSI). Good general infection control practices (e.g. hand hygiene, cleaning and disinfection) are important along with specific measures pertaining to surgery.

Surgical Theatre

The surgery suite should only be used for surgical procedures. It should be isolated from through traffic, ideally in a separate room with entry restricted to critical personnel trained in operating room procedures.

- + Easy to clean and disinfect
- + The surgical room should NOT be used for any other purpose between surgeries

Personal Protective Equipment

All personnel in the surgery should wear designated scrubs, face (mouth and nose) mask, and cap. Personnel involved in the procedure should also wear a sterile gown and gloves. Where Q fever or swine brucellosis are suspected, additional precautions are required such as face shields, disposable drapes and gowns, and upgraded surgical masks.



Disposable shoe covers

Hand Hygiene

A 5-minute surgical hand scrub should be performed prior to application of sterile gown and gloves. An alternative is to use a commercial combination alcohol-chlorhexidine solution application after thorough hand washing (see Section 1 for details).

Pre-operative care

Preparation of the patient prior to surgery in a room external to the surgery suite is mandatory. Clipping (not shaving) should be performed immediately prior to surgery as shaving has been shown to increase incidence of SSI. All animals undergoing an elective major surgical procedure should be bathed in a medicated chlorhexidine shampoo the day prior to surgery.

Skin preparation

A wide area is clipped then prepared in the following sequence: clean and scrub with an antibacterial soap; treat area with an application of alcohol solution; flood with iodine or chlorhexidine solution. Refillable containers used to hold swabs of all disinfectant solutions can become contaminated with resistant bacteria. Thus, they should be cleaned, disinfected and dried prior to refilling, and the swabs removed from them should NEVER be squeezed so that excess solution returns to the bulk container.



IV catheter placement

(Photo courtesy of Dr. Bronwyn Walker)

PRACTICAL TIP:

Contaminated scrub solutions are a potential vehicle of infections. The same surgical skin preparation approach should also apply to routine IV catheter placement in patients.

Post-operative care

The surgical incision is highly susceptible to post-operative invasion with opportunistic pathogens. Bandaging for 24 hours post-operatively is recommended, along with implementing methods to prevent self-trauma until sutures are removed.

Prophylactic antimicrobial use

The aim of using prophylactic antimicrobial agents is to reduce the incidence of post-operative infection whilst minimising the impact on both the skin and gut microbiome. There is little clear evidence for their use in clean procedures. The period of greatest risk for surgical site contamination is during the surgery itself. If antimicrobials are to be used then administration times and doses should be such that therapeutic levels are likely to be present at the surgery site at the time of incision and for the duration of the procedure. These parameters may vary according to the drug used, the bacterial species of foremost consideration, and the surgery site in question. Current practice is to administer parenterally one hour prior to surgery with a repeat dose administered intra-operatively if the surgical time exceeds two half-lives of the antibiotic.

Moving forward, evidence based studies are required in veterinary science to confirm validity of perioperative antibiotic use.

Instrument sterilisation

Complete sterilisation of all instruments and any item that may contact the surgical field is critical to prevent tissue contamination. Autoclaving (steam sterilisation) is the most common method. Sterility indicator strips should be placed in every surgical pack, as the indicator tape is not indicative of a pack's internal sterility.

A biological sterility assay should be used periodically (weekly) or after any autoclave repair.

The *Bacillus stearothermophilus* spore test is the most common commercially-available assay. Spore deactivation during sterilisation is indicated by inability to grow in a suitable growth medium, with a colour reagent indicating bacterial growth after 8-72 hours incubation (depending on the test) following the sterilisation cycle.

Flash sterilisation should only be used for emergency use and never for surgical implants.

Cold sterilisation using immersion in antiseptic solutions

Whilst this can be an effective method, misuse will lead to inadequate sterilisation. Iodophors, alcohol, phenolics and most quaternary ammonium compounds are NOT completely effective as sterilisers and should not be used on instruments intended for invasive procedures. Only glutaraldehyde and stabilised hydrogen-peroxide solutions are effective, but only on clean instruments, in correct and properly maintained solutions with a prolonged (10 hour) contact time. Many solutions are tissue irritating and must be thoroughly rinsed with sterile saline/water prior to use. Instruments left soaking in an iodophor or chlorhexidine solution are NOT sterile and should not be used for invasive procedures.

COMMONLY PERFORMED HIGH RISK PROCEDURES

There are many steps that need to be taken to ensure good infection control practice is followed in everyday procedures that carry a higher risk of organism transmission between patients and hospital personnel. Whilst the following list is not exhaustive, it covers procedures agreed by AIDAP that either represent the highest risk of nosocomial transmission or are often overlooked in an infection-control programme.

A. OTOSCOPIC EXAMINATION IN A CONSULT ROOM

There is a high probability of bacterial contamination of hands, otoscope cones and the otoscope itself during this procedure. Thus very good hygiene needs to be followed after the examination.

Follow the routine hand hygiene and personal protective equipment guidelines as outlined in Sections 1 & 3.

Instrument sterilisation

Otoscope handpiece

It is not possible to autoclave or completely immerse this in any sterilising solution. Thus it should be cleaned in the following manner:

- + Wipe the handpiece of the otoscope with a soapy wet cloth to remove visible debris.
- + If not available then clean thoroughly with a disinfective solution (e.g. alcohol, formaldehyde, bleach).
- + Wrap the handle in non-adherent bandage (e.g. Vetwrap®) or enclose in a disposable rubber glove prior to use to minimise heavy contamination; this should never substitute for thorough cleaning.



Autoclaved otoscope cones

(Photo courtesy of Dr. Mike Shipstone)

Otoscope Cones

Several studies have demonstrated the ineffectiveness of most of the cold soak solutions commonly used in practice to clean otoscope cones. Cones sitting in solutions can breed organisms like *Pseudomonas spp*. Thorough cleaning, disinfecting and drying of cones after SINGLE USE is highly important.

- + All cones should be cleaned after a SINGLE USE.
- + In the case of unilateral disease; examine the unaffected ear first.
- + Change cones between ears even if the ears appear normal.
- + Cones should be autoclaved for cleaning.
- + A practical way of achieving this is to have 10 to 20 otoscope cones and package them up for the autoclave at the end of the day.

B. EAR FLUSHING

Ideally, an ear flush should be performed under a general anaesthetic as it permits debris to be suctioned from the very distal end of the canal, complete examination of the tympanic membrane and irrigation of the middle ear (if necessary). Attempting an ear flush under sedation, will generally only effectively clean the vertical canal and proximal half of the horizontal canals. It is common for patients with chronic otitis externa to harbour very high bacterial or yeast counts some of which may be multidrug-resistant organisms. Great care should be taken to avoid spread of such organisms.



Setting up for an ear flush

(Photo courtesy of Dr. Mike Shipstone)

Procedures area

The ear flush procedure will produce relatively large volumes of potentially highly-contaminated fluid. As such it should be performed on a wet bench, where any fluid can drain and be collected in a sink below the animals head. If this is not available then absorbent pads should be placed under the head and neck.

This procedure should not be performed in the same site as sterile surgical procedures.

Animal preparation

The animal is placed in lateral recumbency with the affected side up. The concave aspect of the pinna is surgically clipped along with an area below the pinna on the lateral neck. This prevents accumulation of any debris and excess medication and matting of the coat.

A small hole is cut in the middle of an absorbent pad and this is placed so that the affected pinna protrudes through. As the ear is gently flushed with saline, the pad absorbs the majority of the drain solution minimising contamination of the wider coat and environment.

Personal Protective Equipment

All personnel should wear designated scrubs, gloves and disposable gowns.

Instrument sterilisation

Clippers

As the hair that has been clipped has a very high risk of serious bacterial contamination with yeast i.e. *Malassezia pachydermatidis* or multi-resistant organisms, the clippers and clipper blades need to be thoroughly cleaned and disinfected after use.

The body of the clipper should be wiped with a soapy wet cloth to remove any visible debris. The body should be thoroughly cleaned with a disinfection solution (e.g. alcohol/chlorhexidine, bleach).

A stiff brush should be used to remove all visible debris from the blade and can then be sterilised using a cold steriliser (e.g. glutaraldehyde), or by autoclaving.

ET Tubes

If the tympanic membrane is ruptured, flushing the external ear canal will lead to contaminated fluid passing down the auditory (Eustachian) tube and into the pharynx. As a result, the ET tubes will be contaminated and require a high level of disinfection. The normal cleaning protocol is insufficient and all ET tubes should be soaked in a cold sterilising solution for adequate time, prior to being rinsed and dried before subsequent use (see Page 24 Anaesthetic Equipment Disinfection and Page 27 Cold Sterilisation using Immersion in Antiseptic Solutions).

C. DENTAL PROCEDURES

It is common for the oral cavity, teeth and gingival pockets to be inhabited with extremely high bacterial counts. Many of these organisms produce biofilms, making antimicrobial control challenging. The use of prophylactic antibiotics for dental procedures is controversial, and antibiotics should only be used as an adjunct to mechanical debridement.

Procedures area

Dental procedures have the potential to produce large volumes of highly contaminated fluid that can be aerosolised with either the drill head, irrigator or scaling instruments. These procedures should be performed on a wet bench, where fluid can drain and be collected in a sink below the animal's head. If this is not available, then absorbent pads should be placed under the head and neck. Dental procedures should not be performed in the same site as sterile surgical procedures.

Animal preparation

The animal is placed in lateral recumbency. The hair may be clipped from the lip margin (depending on the procedure). In cases where heavy bacterial contamination is likely, the mouth (including gingival pockets) should be first flushed with a 0.2% chlorhexidine solution.

Personal Protective Equipment

As there is a high risk of aerosolisation of highly contaminated material, all personnel should wear designated scrubs, gloves, mask (or full facemask) and disposable gowns.

Instrument sterilisation

As above for information on clippers.

Dental Instruments

Handheld

- + Cleaned thoroughly to remove any surface material and then autoclave.

Drill head /Irrigator/Scaling handpiece

- + Some models can be autoclaved and this is the ideal sterilisation method.
- + If it is not possible to autoclave, then instruments should be thoroughly cleaned and disinfected because of the increased risk of serious contamination and transmission of diseases between patients e.g. FIV, FeLV.
- + All instruments should be soaked in a cold sterilising solution until cleaning takes place to prevent any drying of organic material on the surface; dismantled and thoroughly cleaned with a soapy solution, including any fluid channels.
- + Soak instruments in a cold sterilising solution according to the individual manufacturer's recommendations (e.g. glutaraldehyde)
- + Thoroughly rinse and dry prior to subsequent use.



Dental equipment

(Photo courtesy of Dr. B Walker)

ET Tubes

Contaminated fluid is produced with dental procedures with a high probability of passing into the pharynx. As a result, the ET tubes will be contaminated and require a high level of disinfection. All ET tubes should be soaked in a cold sterilising solution for the prescribed time; rinsed and dried before subsequent use (see Page 24 Anaesthetic Equipment Disinfection and Page 27 Cold Sterilisation using Immersion in Antiseptic Solutions).

SECTION 5: BIOSECURITY, INFECTION CONTROL AND PREVENTION OF COMMON ZOONOTIC INFECTIONS

This section provides important information on possible risks associated with infectious disease agents that veterinarians who work predominantly in companion animal practices in Australia may encounter. It is important that veterinarians make themselves aware of these agents and the threats that these may impose for the health of their staff.

ANIMAL BITES

A large number of commensal organisms of the mouth of dogs and cats can cause opportunistic infections in humans following a traumatic bite wound. In one study, approximately 20% of bite wounds became infected, with cat bite wounds three times more likely to become infected compared to dog bite wounds.

- ✚ The most common organisms isolated from bite wound infections are *Pasteurella multocida*, *Capnocytophaga* spp., *Streptococcus* spp. and Gram-negative anaerobic bacteria. Incubation periods are typically short, requiring prompt attention to the wound inflicted.
- ✚ All bite wounds that puncture the skin should be immediately and thoroughly irrigated, and cleaned with antiseptic solution i.e. chlorhexidine or iodine.
- ✚ For bite wounds with a delayed presentation, puncture wounds that cannot be debrided, wounds on the hands, feet or face and/or those involving deeper tissues such as tendons, bone and joints, prophylactic antimicrobial chemotherapy is indicated.

In a veterinary setting, clinicians, especially hospital owners and superintendents, are strongly recommended to have their staff treated as hospital outpatients or by a local medical practitioner. In severe cases, or if the victim is immunocompromised, it is prudent to request also an infectious disease consultation. In especially severe cases, hospitalisation and treatment with intravenous antimicrobial therapy is required.

BRUCELLOSIS

Brucellosis is an uncommon disease in Australia. However, there are medium to high-risk groups in the population (feral pig hunters and their dogs; dogs fed feral pig meat; and staff in veterinary hospitals and pathology laboratories).

Brucellosis is a notifiable disease in humans and animals in some states of Australia. Since eradication of *Brucella abortus* in cattle and buffalo in Australia, very few human brucellosis cases have been reported. *Brucella suis*, is a much more serious zoonotic pathogen than *B. abortus* and is endemic in the feral pig populations of Queensland, northern New South Wales and parts of Western Australia and the Northern Territory.

- + In humans in Australia, approximately 10-50 cases of *B. suis* infection are diagnosed annually, with most cases occurring in pig hunters. Clinical signs in humans are highly variable and may be chronic in nature following dissemination and sequestration of the organism in bone, joints, brain or genitourinary system without prompt effective treatment *

Several recent cases of *B. suis* infection have been reported in pig hunting dog breeds in northern New South Wales and Queensland with the main presenting signs being fever, malaise, back pain and shifting lameness from associated discospondylitis or orchitis/epididymitis. Humans and dogs may acquire *B. suis* infection during hunting and carcass dressing from either contamination of cuts and abrasions, or exposure to aerosols. Consumption of undercooked pig meat is also an important risk factor, especially for dogs. Laboratory staff may also be at risk of infection as the organism aerosolises off the plate, requiring work to be performed in a biosafety cabinet at all times.



PRACTICAL TIP:

Brucellosis due to *B. suis* should be considered in the differential diagnosis for breeds of dog presenting with clinical signs of fever and spinal pain (discospondylitis) or enlargement of the testes. Neutered animals tend to develop discospondylitis, while sexually intact animals develop either orchitis/epididymitis (males) or abortion (females).

Orchitis and epididymitis in a dog with *Brucella suis* infection.

This photograph was obtained intraoperatively, after 2 weeks of preliminary therapy with doxycycline and rifampicin.
(Photo courtesy of Dr. Richard Malik)

Dogs considered at high risk should be serum sampled for serological testing (available through most Department of Primary Industries Veterinary Diagnostic Laboratories [VDL]) prior to any surgical procedure, such as castration, being performed. **Culturing *Brucella* from infected clinical material is hazardous for laboratory staff and they should be well informed of this possibility prior to submission of any suspect samples (blood, urine, tissues i.e. testes). Humans inadvertently exposed to contaminated tissues should consider prophylactic antimicrobial treatment.**

Personal protective equipment including disposable gowns, gloves, surgical mask and eye protection should be mandatory for all surgical staff if a procedure is going to be performed on a confirmed sero-positive dog. Only 10-100 organisms are needed to constitute an infectious aerosol for humans. Thorough cleaning and disinfection of the surgical suite should be carried out wearing appropriate PPE following the surgical procedure. Specimens should be aseptically collected in leak-proof sample containers with adequate notification of their potential risk prior to submission to a VDL.

*Abrupt onset of fever (persistent, intermittent or irregular), chills and sweating may be accompanied by myalgia/arthalgia, headaches, and mental depression. Lymphadenopathy, hepatomegaly and splenomegaly may accompany infection or signs may be vague and non-specific.

- + Great care should be taken when handling aborted foetuses from a potentially infected bitch. Treatment using doxycycline plus rifampicin for 2 weeks is recommended, and then the affected testis or testes should be removed surgically.
- + Surgeons should use protective glasses, masks, disposable gowns and gloves. The zoonotic risk would appear to be lowered by using the preceding antibiotic therapy for 2 weeks.
- + It is prudent to continue combination therapy for 4-8 weeks following surgery, while cases with discospondylitis require even lengthier treatment regimens.

LEPTOSPIROSIS

In areas where leptospirosis is endemic, it should be suspected in dogs presenting with lethargy, inappetence, fever, jaundice, vomiting, diarrhoea and renal failure. The classic presentation for *Leptospira interrogans* serovar *australis* in far north Queensland is sudden illness with red/brown urine and jaundice. A mortality rate of over 50% occurs in dogs progressing to disseminated intravascular coagulation. Demonstration of a high titre (1:400 or greater) or a four-fold rise in specific antibody titre in paired serum samples in the microscopic agglutination test (MAT) is confirmatory. Confirmation of diagnosis by PCR on blood or urine specimens should also be attempted. As testing results may take time to return, if leptospirosis is suspected, treatment in the form of oral doxycycline for 7 days should be commenced immediately (no later than the fourth day after onset) whilst awaiting the results of diagnostic testing. Severe leptospiral infection requiring hospitalisation is usually treated with IV penicillin.

Leptospirosis is a notifiable disease in humans and animals in Australia.

Specific *Leptospira* serovars are associated with maintenance in reservoir hosts and frequent shedding of the organisms in urine. In incidental hosts, in which infection is often more severe and involving multiple organs, shedding in urine is not typically a feature. A large number of serovars have been identified in Australia, mainly occurring in tropical and subtropical Queensland and the Northern Territory, and dogs and humans are often accidental hosts of more virulent serovars, including *copenhageni*, *grippotyphosa*, *zанони* and *australis*.

- + Infections with most serovars are sporadic and few cases have been documented in companion animals. In contrast, infections with *L. australis* are common and seasonal in places like Bowen, Cairns and Townsville.
- + The maintenance hosts for these serovars are usually rodents (introduced and native) and small marsupials, while infections in both humans and dogs occur most frequently following heavy rainfall and warm temperatures.
- + The main potentially zoonotic *Leptospira* serovar for which dogs are the maintenance host (*canicola*) and thus are likely to frequently shed live leptospires in their urine, is not present in Australia.
- + Cats are relatively resistant to *Leptospira* infection, although seroconversion occurs.

Further studies are required to determine the true zoonotic risk of dogs and cats with leptospirosis. In the majority of infections occurring in Australia, dogs are considered to be incidental hosts and the period of shedding of live leptospires in urine is likely to be short. Whilst clinically ill animals with leptospirosis are likely to represent minimal risk of zoonotic infection to in contact humans, precautions should be taken.

PRACTICAL TIP:

Animals should be kept in isolation and urine should be disposed of safely and disinfection/disposal of soiled articles undertaken. Clinically ill animals should be examined wearing disposable gowns, gloves and eye protection. Avoid contact of open wounds or abrasions with urine and clean and wash exposed skin thoroughly after handling the animal.

PSITTACOSIS

Psittacosis is caused by *Chlamydia psittaci* and infection is particularly common in psittacine birds including parakeets, cockatiels, budgerigars and lovebirds. Affected birds develop ocular nasal discharge (with sneezing), conjunctivitis and systemic infection characterised by diarrhoea with green-tinged urates. Latent infections may be reactivated by stress such as introduction of a new bird to an aviary. Large numbers of chlamydia are shed in the respiratory secretions and faeces of clinically ill birds which are highly infectious.

PRACTICAL TIP:

Psittacosis should be suspected in any bird that has non-specific signs of illness, especially if it has been recently acquired or stressed.

Infection of humans typically results from inhalation of aerosols following prolonged and close contact with infected birds, cleaning of aviaries and cages, or performing necropsy examinations of dead birds without appropriate personal protective equipment (PPE). However, in many cases, exposure is incidental and cannot be attributed to known bird contact. Infection in humans can range from a mild, non-specific, flu-like illness to life threatening pneumonia. Multi-organ systemic infection is a rare sequelae. Approximately 100 cases of psittacosis occur in humans in Australia per annum.

Given the high prevalence of psittacosis in caged birds, information leaflets on psittacosis prevention should be provided to clientele who are aviarists or bird fanciers, including use of PPE (disposable overalls, P2 particulate filtering face mask, and gloves) when cleaning out aviaries, quarantine of introduced stock, and directions to seek prompt medical advice if they experience flu-like symptoms following an exposure event. Based on history, the receptionist should provide instructions that birds presenting to the hospital with overt clinical signs should not be admitted to the waiting room, rather the owner should remain outside until called through at the time of their appointment with the veterinarian.

- ✚ Clinical examination and diagnostic sampling of live birds can be undertaken wearing a face mask, making sure the examination is undertaken in a room with no air conditioner draughts or ensuring handlers place themselves upwind (i.e. between the bird and draught source) of the air flow.
- ✚ Hospitalised birds with suspected chlamydiosis should be placed in isolation with the above precautions taken into account during triage and cage cleaning.
- ✚ *C. psittaci* is susceptible to most disinfectants; however, it is resistant to acid and alkali.
- ✚ Necropsy examinations should be performed in a biosafety cabinet, or if this is unavailable, the post-mortem can be performed on a surgical table in a draught-free room wearing full PPE. Feathers should be well wetted with 70% ethanol.

Owners who have had intimate contact with birds diagnosed with chlamydiosis should be advised to seek medical advice immediately if they develop flu-like symptoms over the next 14 days. Diagnosis of chlamydiosis in birds is typically based on a combination of antigen or PCR detection from swabs of ocular nasal discharge and faeces, and high antibody titre in serum samples correlated with high white cell count and/or liver enzymes on ancillary blood tests. The incubation period for psittacosis in humans is typically 5-14 days.

Diagnosis of psittacosis in humans can be problematic as sputum or blood samples are often negative by PCR. Serological diagnosis for specific IgG response is a more reliable indication of infection but requires four-fold or greater increase in antibody titre in paired samples between acute and convalescent phase serum samples or a high titre from a single acute phase sample. Complications have arisen in patients presenting with acute *C. psittaci* pneumonia who are treated with inappropriate antimicrobials normally prescribed for community-acquired pneumonia and continue to deteriorate.

As in birds, the treatment of choice for *C. psittaci* infection in humans is doxycycline. A rapid response to treatment usually ensues. A history should be provided to the attending physician indicating recent exposure to birds to ensure correct diagnostic testing and antimicrobial choices are made.

Q-FEVER

Exposure to peri-parturient breeding cats and dogs is a significant risk for development of coxiellosis. Q-fever is the disease in humans caused by the bacteria *Coxiella burnetii*. A large range of animal species may be infected, most asymptotically, with the organism most commonly associated with ruminants and wildlife. Several hundred cases are reported in humans each year, most commonly occurring in northwest NSW and central/south west Queensland.

Q-fever outbreaks have occurred in veterinary staff in a veterinary clinic associated with peri-parturient animals of both dogs and cats; the most high-risk veterinary procedure for exposure to *Coxiella burnetii* in companion animal practice is likely to be a caesarean section or other obstetrical interventions. Veterinary hospitals with a large number of cat breeding clients could consider serological testing of breeding queens to confirm exposure and likely risk.

All personnel attending the surgery should wear disposable gowns over surgical scrubs, and gloves, surgical masks and disposable drapes should be used. Avoid creating aerosols during surgery and intimate contact with delivered kittens (i.e. mouth-to-mouth or mouth-to-nose resuscitation). *C. burnetii* is resistant to many disinfectants including sodium hypochlorite. Following surgery, all contaminated disposable materials should be aseptically placed in an autoclave bag and sterilised. Following cleaning, 70% ethanol spray should be used on all exposed surfaces and wiped clean before removing PPE. Care must be taken to dispose of all fabrics used to wrap and resuscitate kittens as standard washing and drying (clothes drier or air drying) may facilitate aerosol formation and organism transmission.

PRACTICAL TIP:

When performing caesarean sections on breeding queens, ensure that all personnel working in the vicinity are either vaccinated against Q-fever or have evidence of previous exposure. Exclude other personnel and owners.

Q-fever is a notifiable disease.

The incubation period in humans ranges from 4 days to 6 weeks, but is typically 2-3 weeks. Infection is asymptomatic in 60% of cases. Clinical symptoms resemble a typical “flu-like” illness (lasting 2-6 weeks) with fever, headache, sweats, fatigue and anorexia/weight loss[†]. Chronic disease manifestation and even chronic fatigue-like syndrome can occur in anyone infected and does not have to be preceded by overt acute flu-like symptoms.

[†]More serious acute infections include pneumonia and hepatitis. Chronic infections include myocarditis which can be fatal but is rare (1-2% of cases) and a post-infection chronic fatigue-like syndrome (10-20% of cases).

To avoid possible long-term complications, treatment should be administered as early as possible and consists of doxycycline for at least 14 days' duration. Q-fever vaccination is recommended for those at high risk of exposure, including veterinarians, veterinary nurses and other employees with high animal contact, particularly with peri-parturient animals. In a recent survey, 89% of Australian veterinarians were vaccinated, compared to only 29% of veterinary nurses. Unvaccinated individuals must first be tested for Q-fever antibodies as well as an intradermal skin test for delayed type hypersensitivity prior to Q-fever vaccination. Individuals with immunological evidence of prior exposure (high antibody titre and/or positive skin test) should not be vaccinated (as they may develop symptomatology consistent with chronic Q-fever) and Q-fever vaccination and/or testing is not recommended for individuals under 15 years or during pregnancy.

OTHER IMPORTANT DISEASES WITH ZOONOTIC POTENTIAL

Additional diseases listed below are potentially zoonotic. In each situation, if exposure or contact is deemed likely, protective precautions should be undertaken. These are listed below.

Dermatophytes (*Microsporum canis* 'ringworm')



Child with facial ringworm lesions

(image courtesy of Dr. Richard Malik)

The main risk would appear to be transfer of material from contact with cats, especially infected kittens. There is a high rate of carriage of dermatophytes in cats, especially long-haired Persian lines, with *Microsporum canis* being particularly common. Carriage is more common in young cats and cats from multi-cat premises and may be asymptomatic. Infections in humans with compromised immune systems or children may be particularly severe. In such cases, care should be undertaken to avoid contact with cats. Fungal culture of feline hair or hair brushings may be used to determine if carriage is present. *M. canis* has been isolated from the floors, examination rooms and wards of veterinary hospitals. Appropriate daily disinfection procedures should reduce the risk of exposure and transmission. In suspected feline cases, gloves should be worn during examination and a protective disposable gown placed over scrub tops to prevent transmission to humans.

Disseminated fungal infections

Many disseminated fungal infections caused by dimorphic fungi have zoonotic potential. Whilst disseminated infections caused by these pathogens are extremely rare in animals in Australia, they have been reported in dogs imported into Australia from the United States (e.g. *Coccidioides immitis*-reactivation of latent infection presumably due to immunosuppression) as well as the occasional sporadic endemic infection in individual dogs and cats (e.g. histoplasmosis, sporotrichosis). Zoonotic transmission from infected animals is either extremely rare or not a significant feature of these systemic mycoses. However, an Australian veterinarian was reported to have acquired systemic sporotrichosis from a non-healing wound infection in a cat.

In direct contrast, *in vitro* culture of many dimorphic fungi can be extremely hazardous to laboratory staff. As the mould form on typical fungal media at room temperature, they form masses of aerial hyphae and spores which easily aerosolise and are highly infectious. Culture must therefore only be attempted in a biosafety cabinet in a certified PC-2 or PC-3 laboratory.

Veterinarians should submit representative fresh tissue biopsies (never swabs) to veterinary diagnostic laboratories (VDL). Diagnosis should be augmented by histopathological examination of formalin-fixed clinical samples and specific PCR tests. Definitive diagnosis on disseminated fungal infections of animals can be provided by mycology reference laboratories around Australia such as The National Mycology Reference Centre based in Adelaide, SA and ICPMR at Westmead Hospital in Sydney, NSW*. It is strongly recommended that VDLs forward cultures to such reference laboratories for species identification and undertake susceptibility testing where appropriate.

PRACTICAL TIP:

If you suspect disseminated fungal infection in an animal due to one of the rare dimorphic fungi (e.g. in a dog or cat previously imported into Australia), notify your VDL and provide as much history as possible with your submission so the appropriate biosafety conditions can be maintained to prevent the risk of laboratory-acquired infection. Always submit fresh tissue specimens (e.g. in sterile urine or wrapped in a saline-soaked gauze swab and formalin fixed tissue biopsies, never swabs).

Campylobacteriosis and Salmonellosis

Animals fed raw meat, e.g. chicken, typically shed infectious organisms in faeces and transmission to people can occur via the faecal-oral route if people fail to observe hand hygiene. Transmission to humans with subsequent clinical signs of gastroenteritis may occur. It is important that animals suspected to be infected with these organisms are managed to prevent human contact. Isolation ward procedures as outlined previously should be undertaken. Gloves and protective clothing are necessary and hand washing after handling is essential. Care with disinfection of fomites such as bedding, food bowls, clothing and food covers is required. Detergents and use of hypochlorite and/or other disinfectants is sufficient for elimination of these organisms.

Bartonella henselae (cat scratch fever)



Precautions for avoiding transmission of *Bartonella* from pets to people include maintaining good flea control. Stray cats and cats less than 1 year of age have been suggested to be more likely to transmit infection, and the flea is the central critical agent responsible for disease transmission through flea faeces, which can easily contaminate scratch injuries. This agent is typically transferred via the nails of infected cats, however a cat licking open wounds on a human may represent a possible route of infection. Cat scratch wounds should be immediately treated with washing in medicated hand washes containing chlorhexidine. Appropriate antibiotic therapy should be implemented if clinical signs of infection develop. These include fever, lymphadenomegaly, erythema or swelling at the site of the scratch.

Multidrug-resistant *Staphylococcus* spp.

Methicillin-resistant *Staphylococcus aureus* (MRSA) and *Staphylococcus pseudintermedius* (MRSP) may be identified in the course of diagnosing and treating infections in dogs and to a lesser extent cats. Such infections represent a source of transmission of an infectious agent to both humans and veterinary patients. Such infections may be symptomatic or asymptomatic but may be very difficult or impossible to treat solely with systemically administered antibiotics. In animals identified with MRSP or MRSA, it is very important that they not be allowed to transmit the bacteria to other animals or humans in the hospital. Animals should be isolated and prevented from direct contact with other patients. They should not be moved through the hospital, but if this is necessary they should be transported on a gurney covered with disposable drapes. Barrier nursing should be implemented and PPE including disposable gloves, gowns, masks and other protective equipment should be used. These materials should be disposed of following use. Thorough cleaning of their entire cage including walls, locks, handles etc is essential to avoid cross contamination. Laundry items should also be washed separately using disinfectant solutions.

Further detailed information is available at:

<https://www.zoetisus.com/Conditions/Pages/Dermatology/education-library/InfectionControl-VetHospitals.pdf>



**Methicillin resistant
Staphylococcus
*pseudintermedius***

*These specialist reference laboratories will only provide definitive diagnosis on in-vitro grown cultures and susceptibility testing, NOT primary culture from diagnostic specimens.

Cryptococcus neoformans/gattii

Infected cats and dogs with cryptococcosis pose no public health consideration to humans. The organism does not aerosolise (to the infectious form of the disease) from sites of tissue infection. Direct inoculation of yeast tissue into the body (e.g. wounds) is a possible route of infection and in such situations precautions should be undertaken such as wearing gloves during surgical biopsy of tissues.

Rabies and bat lyssavirus

Rabies remains an exotic disease in Australia, but the possibility of entrance to the country should not be discounted. In countries where rabies is endemic, exposure of humans is usually related to animal bites from infected animals. In Australia, the related Bat lyssavirus is also potentially zoonotic. Virological and/or serological evidence of ABLV infection has been found in all four species of flying foxes (megachiropterans) found in Australia, and at least seven genera of Australian insectivorous bats. In particular, humans examining both live and dead bats are at risk of exposure to the virus. Four human deaths have been documented following bat exposures in Europe, all presented with clinical features of rabies. Any suspected infected animals should be isolated from other animals and humans, and veterinary investigation/management sought. Where exposure to domestic dogs, cats and ferrets (but not other animals) occurs overseas and where observation of the suspected animal is possible, information on whether the dog, cat or ferret remained healthy for at least 10 days after the exposure incident may be useful for assessing risk of infection and the need for completion of post exposure prophylaxis.

Environmental contamination by infected animals is considered negligible; this is based on knowledge of persistence of the classical rabies virus, which is fragile and does not survive for long outside the host. It is readily inactivated by heat and direct sunlight. Bats or other animals that have been dead for longer than 4 hours are no longer considered infectious for lyssaviruses. Bat or other animal blood, urine, and faeces are not considered to be infectious. Post mortem examination of tissues from dead bats or examination of animals possibly exposed to the Bat lyssavirus should be undertaken with extreme care. Consultation with animal health colleagues is strongly recommended. Typically post mortems of suspected animals should only be conducted in facilities designed to prevent human exposure to aerosols.

Further information is available at:

<http://www.health.gov.au/internet/main/publishing.nsf/Content/cdna-song-abvl-rabies.htm#toc10>

Giardiasis

It has been shown that genotypes representing *Giardia* isolates from dogs and cats have some but limited potential to be zoonotic. The genetic typing of *Giardia* is required to determine if there is a potential for the *Giardia* strain to be regarded as zoonotic. In view of this, most veterinarians should consider any dog or cat infected with *Giardia* to be potentially zoonotic. Appropriate procedures to prevent transmission should be undertaken as described previously for *Salmonella* and *Campylobacter* infections.

Toxoplasmosis

Cats are the definitive host of *Toxoplasma gondii* and become infected by eating infected rodents, birds, or other small animals. The parasite is then passed in the cat's faeces in an oocyst form, which then sporulates and becomes infectious. Kittens and cats primarily infected with *Toxoplasma* can shed oocysts for around 3 weeks after infection. Mature cats are less likely to shed *Toxoplasma* if they have been previously infected. A *Toxoplasma* infected cat shedding the parasite in its faeces is a potential source for human transmission via the faecal-oral route. The following are suggested preventative measures for preventing cat to human transmission in veterinary hospitals.

Collect and dispose of all cat litter/faecal matter daily. The *Toxoplasma* parasite does not become infectious until 1 to 5 days after it is shed in a cat's faeces. Protective gloves are to be used to handle litter boxes and cat faecal contaminated bedding or cages. The risk of transmission to pregnant women and subsequent development of foetal infections is the most serious consequence of zoonotic infection with *Toxoplasma*. Pregnant women or potentially pregnant women should take strict precautions to avoid contact with cat faeces.

FINAL NOTE

This is meant to be a 'living document'. Hospital infection control guidelines are likely to change over time as new products became available, and as new disease entities emerge, even though the underlying principles remain the same. When confronted with new infection control challenges, it is worth performing key word searches to keep abreast of on-going developments in this dynamic field.

AUSTRALASIAN INFECTIOUS DISEASES ADVISORY PANEL



Dr Steve Holloway

BVSc (Syd), MVS, PhD (Melb), DACVIM, MACVSc

Steve graduated from the University of Sydney in 1983 and after several years in practice, journeyed along the pathway to becoming a specialist in internal medicine. In 1991 Steve became a Diplomate of the American College of Internal Medicine. In 1994 Steven commenced his PhD studying herpesviruses of horses at the University of Melbourne, completing in early 1998. Between 1999 and 2009 Steve lectured in infectious diseases of small animals at the Faculty of Veterinary Science at the University of Melbourne. Steven is currently a specialist in internal medicine in private practice and regularly consults on infectious disease problems in small animal patients.



Professor Darren Trott

BSc (Hon), BVMS (Hon), PhD

Professor Darren Trott is a veterinarian with 25 year's experience in bacterial disease research focused on zoonotic infections, enteric diseases, gastrointestinal microbial ecology and antibiotic resistance. In 2010, Darren accepted a position in the new School of Animal and Veterinary Sciences at The University of Adelaide and is Director of the Australian Centre for Antimicrobial Resistance Ecology, a new Research Centre focused on antimicrobial resistance surveillance and finding One-Health solutions for multidrug-resistant pathogens in both animals and humans.



Dr Mike Shipstone

BVSc, FACVSc, DACVD

Dr Shipstone graduated from Queensland University in 1984 and has worked in a number of different private practice and industry positions. In 1995 he started a residency at the Animal Skin and Allergy Clinic in Melbourne, with additional periods of study at the University of California, Davis and Louisiana State University, Baton Rouge. Mike is principal and director of a specialist dermatology referral practice and adjunct Associate Professor at the University of Queensland. Mike is a Fellow of the Australian College of Veterinary Scientists (Veterinary Dermatology) and a Diplomate of the American College of Veterinary Dermatology, the only dual boarded veterinary dermatologist in Australia. Mike has published in Australia and overseas and has presented in Australia, South East Asia and North America.



Professor Vanessa Barrs

BVSc (hons), PhD, MVetClinStud, FANZCVS (Feline Medicine), GradCertEd (Higher Ed)

Professor Vanessa Barrs is the Director of the University Veterinary Teaching Hospital. She established specialist veterinary services and clinical research at the Valentine Charlton Cat Centre in 2004 and is a registered Specialist in Feline Medicine. Professor Barrs' research group investigates companion animal infectious diseases, especially fungal and viral infections. Vanessa was awarded Distinguished Scientific Award in 2009 for her research by the Australian Small Animal Veterinary Association. She has over 100 refereed publications and book chapters.



Dr Richard Malik

DVSc, DipVetAn, MVetClinStud, PhD, FACVSc, FASM

Richard Malik graduated from the University of Sydney, trained in Anaesthesia and Intensive Care, and then moved to ANU where he completed a PhD in pharmacology at the John Curtin School of Medical Research. He then completed a Postdoctoral fellowship at the Neurobiology Research Centre before returning to his alma mater where he remained there for 16 years in a variety of positions (1995 to 2002). Since 2003 Richard has worked as a consultant for the Centre of Veterinary Education and he finds time also to see cases in a number of practices in the Eastern suburbs of Sydney. Richard has varied research interests, most notably infectious diseases, genetic diseases and diseases of cats in general. He is a Fellow of the Australian Society of Microbiology, a member of the Australian Society of Infectious Diseases and an Adjunct Professor of Veterinary Medicine at Charles Sturt University.

AUSTRALASIAN INFECTIOUS DISEASES ADVISORY PANEL



Dr Mandy Burrows

BSc, BVMS Murd, MACVSc, FACVSc (Dermatology)

Mandy is a Fellow of the Australian College of Veterinary Scientists in Veterinary Dermatology and a registered specialist in veterinary dermatology. She is a consultant in veterinary dermatology and has two dermatology practices in Perth, Western Australia that provide secondary and tertiary referral advice for skin, ear and allergy problems in dogs, cats and horses. She lectures in dermatology at Murdoch University Veterinary Hospital and she teaches undergraduate veterinary students and the dermatology unit of the Masters in Veterinary Medicine at both Murdoch and Massey University, New Zealand. She is currently the Chief Examiner and serves on the Council and the Board of Examiners of the Australian and New Zealand College of Veterinary Scientists and is a member of the Advisory Committee for the Registration of Veterinary Specialists. She is a member of the Australian Advisory Board for Infectious Diseases in companion animals and is the current Australian and New Zealand representative and the Secretary of the World Association for Veterinary Dermatology. She has extensive experience with clinical dermatology in companion animals and she enjoys teaching dermatology to veterinary undergraduate and postgraduate students.



Associate Professor Jacqueline Norris

Associate Professor Jacqueline Norris BVSc MVS, PhD, MASM, MASID Grad Cert Higher Ed.

Jacqui is a veterinarian and veterinary microbiologist, passionate about providing clinically relevant infectious disease courses for veterinary students, practitioners and breeders. She is heavily involved in developing the structure, content and governance of the new Doctor of Veterinary Medicine (DVM) curriculum. Her research includes: Development of diagnostics and treatments for companion animal viral diseases, Q fever, multi-resistant (MDR) *Staphylococcus* species, Chronic Renal Disease in domestic and zoo Felids.



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