THE UNIVERSITY OF QUEENSLAND
AUSTRALIA

School of Veterinary Science

The University of Queensland

Biosecurity, Hygiene and Infection Control Manual

Standard Operating Procedures for UQ-VETS Small Animal Hospital, Clinical Studies Centre, Gatton and UQ-VETS Dayboro

Contributors:

Many staff of the Veterinary Science, University of Queensland School of contributed to sections of the original version of this manual, including, but not limited to (in alphabetical order):

Tamsin Barnes
Cameron Broome
Lisa Bubb
Philip Chamberlain
Trish Clarke
Rowland Cobbold
Anne Covill
Margaret Day
Bob Doneley
Trisha Farry
Anna Galloway
Susan Keane
Myat Kyaw-Tanner
Lisa Kidd
Paul Mills
Annie Rose
Dan Schull
Rebekah Scotney
Andrew Van Eps
Dick (John) Wright
Steven Zedler

The manual was compiled by Dan Schull (d.schull@uq.edu.au) and Ristan Greer (r.greer@uq.edu.au).

In December, 2017 School's Occupational Health and Safety Co-ordinator together with several staff from Veterinary Medical Centre, Gatton have reviewed and revised the original version with reference to http://www.ava.com.au/sites/default/files/Guidelines-for-veterinary-personal-biosecurity-2017-FINAL.pdf.

TABLE OF CONTENTS

INTRODUCTION

Biosecurity and infection control are increasingly important in veterinary practice. All professionals involved in veterinary practice have a responsibility to ensure the safety and welfare of people and animals involved in veterinary care. Infection prevention and control strategies are designed to protect patients, owners, veterinary personnel, students and the community. A significant percentage of hospital-associated infections in veterinary clinics can likely be prevented with proper compliance to basic, practical infection control practices. A systematic approach to infection prevention and control requires all personnel to play an active role in protecting every person and animal associated with the veterinary clinic, patients or veterinary personnel. Veterinary personnel need to follow infection prevention and control protocols at all times and apply critical thinking and problem solving in managing clinical situations.

This practical biosecurity and infection control guideline is intended to ensure that all staff, students and clients who interface with the clinical veterinary services of the School of Veterinary Science, University of Queensland are aware of the minimum standards expected, and that procedures and principles are similar in every location. This guideline is not intended to be a comprehensive document covering all aspects of biosecurity and infection control.

Understanding and application of excellent infection control principles are important requirements for accrediting bodies for veterinary schools. Biosecurity and infection control procedures are evaluated during accreditation visits. In Queensland, veterinary surgeries, hospitals and clinics are required to comply with Occupational Health and Safety requirements relating to infection control as well as other areas.

Overall, the aims of this document are to:

1. Protect staff, students and clients from exposure to zoonotic disease agents
2. Minimise the risk of nosocomial (hospital-acquired) infection to patients
3. Ensure students learn and apply best-practice in biosecurity and infection control in the clinical setting
4. Educate, by example, clients and members of the public in biosecurity and infection control
5. Provide a clean, safe and attractive working environment for everyone concerned
6. Protect operational capabilities of the clinics

To follow, we have included excerpts from two key reference documents used during the development of this Biosecurity, Hygiene and Infection Control Manual:

(1) An excerpt from the 'Australian Veterinary Association Policy for Infection Control' and 'Code of Practice for Management of Hygiene and Infection Control'
(2) A summary of key strategies for 'Infection Prevention and Control Best Practices For Small Animal Veterinary Clinics' developed by the Canadian Committee on Antibiotic Resistance (2008)

AVA policy 3.3 - Code for Infection Control

The Australian Veterinary Association (AVA) supports practices that:

- ensure the safety and welfare of all animals under veterinary care
- provide a safe and healthy working environment for owners, veterinarians and staff

Animal hospitals and veterinary practitioners have a duty of care and must take reasonable action to safeguard animals, staff and the public from infection. Employers must establish procedures and provide information, training and supervision, especially for infection control.

Veterinarians must be conscious of both the potential for zoonoses to present as unapparent infections in animals, and of their responsibilities regarding cross infection amongst animal patients. They must recognise the potential for pathogens to be introduced through inadequate infection control during administration of medication.

The following recommendations are compiled from the 'AVA Code of Practice for Management of Hygiene and Infection Control for Veterinarians':

1. Workplace based infection control plan
   Each workplace, including ambulatory services, attended by the veterinarian will be assessed for the risk of infection to all workers, clients and animals. The risk assessment will include development and audit of standard operational procedures to minimise the risk of adverse consequences from a foreseeable event at that workplace. The risk assessment will be documented and reviewed at least annually against published material for the control of infection in veterinary workplaces. A senior professional staff member should have responsibility for currency of the plan and inclusion of all staff in the development and application of the procedures to manage risks.

2. Standard Infection Control Procedures
   All staff involved in the handling of animals, animal waste or material that may be contaminated with animal fluids or veterinary therapeutic agents will be regularly instructed in the correct use of standard infection control barriers and monitored for correct application of these practices.

3. High Risk Procedures
   Veterinarians who are undertaking high risk procedures, where the risk of adverse consequences would be very likely or very severe due to the nature of the procedure or infectious agent need to ensure additional precautions are observed for themselves and any at risk human or animals. High risk procedures would include procedures where aerosol dispersion or gross contamination by body fluids could occur while undertaking the veterinary procedure. Additional protection levels including exclusion of nonessential persons or animals and the use of specialised protective equipment are required.

4. Demonstrated commitment to infection control practices
   Veterinarians must demonstrate a commitment for continual application of the principles of protection of themselves, and all other human and animals under their control. Verbal instruction and educational material is vital for all staff at their work place. Written material must also be available for clients of high risk animals. Staff are required to actively participate in developing standard procedures for management of risks as well as reporting new risks and incident reporting.

5. Monitoring of infection control programs
   The monitoring and documentation of participation by staff and clients in management and minimisation of risk of infection within the workplace must be encouraged through formal audit procedures. Demonstrated continued professional education programs as well as informal discussions with staff and clients should be documented as evidence of application of the infection control plan and a commitment to minimisation of the risks to all persons and animals.

The following summary for infection prevention and control best practices or small animal veterinary clinics is written by the Canadian Committee on Antibiotic Resistance (2008):

1. Infection prevention and control strategies are designed to protect patients, owners, veterinary personnel and the community. All veterinary personnel should play an active role in protecting every person and animal associated with the veterinary clinic.

2. Every veterinary clinic, regardless of type or size, should have a formal infection control program, a written infection control manual, and an infection control practitioner (ICP) to coordinate the program.

3. Some form of surveillance (either passive or active) should be practiced by all veterinary facilities. The keys to passive surveillance are to centralize the available data, and to have a designated ICP who compiles and evaluates the data on a regular basis.

4. Routine Practices that are critical to infectious disease prevention and control:

a. Hand hygiene, including hand washing and the use of alcohol-based hand sanitizers

b. Risk reduction strategies, particularly those related to: the use of personal protective equipment (PPE), cleaning and disinfection, laundry and waste management

c. Risk assessment of animals and personnel with regard to: disease transmission and disease susceptibility.

d. Education of veterinary personnel, students, animal owners and the public

5. All surgical procedures cause breaks in the normal defensive barriers of the skin or mucous membranes, and therefore carry an inherent risk of surgical site infection (SSI). Good general infection control practices (e.g. hand hygiene, cleaning and disinfection) are important for

prevention of SSIs, but there are also specific infection control measures pertaining to surgery that should be considered.

6. Every veterinary clinic should have an isolation area for caring for and housing animals with potentially contagious infectious diseases.

7. Proper wound care is critical to preventing transmission of bacteria, particularly multidrug-resistant pathogens, between animals, personnel and the environment.

8. Animals from shelters and similar facilities should be considered high risk from an infectious disease standpoint and managed appropriately to prevent transmission of disease.

9. Safety of personnel and animal owners should always be a priority. Personnel should take all necessary precautions to prevent animal-related injuries (e.g. bites, scratches), and all bite wounds should be taken seriously. Proper sharps handling practices should be emphasized to reduce the risk of needle-stick injuries.

10. Education of personnel and clients about zoonotic and infectious disease risks and prevention is crucial.

RATIONALE FOR ROUTINE PRACTICES – THE CHAIN OF TRANSMISSION

Modified from information compiled by the Public Health Agency of Canada, 1999 and Canadian Committee on Antibiotic Resistance (2008)

Transmission of infection during the provision of health care requires three elements:

(1) **A source of infectious microorganisms** – includes animals, people (clothing, hands etc), food, water, medical equip, drugs, bedding, waste material etc. Microorganisms to consider include bacteria, viruses, fungi and parasites. In some cases, vectors such as lice, mosquitoes, flies, ticks, fleas, rodents and other vermin can transmit pathogens.

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

(3) **A means of transmission for the microorganism.** Prevention of infection in animal health care settings should be directed at interrupting the transmission of microorganisms from source to host, because agent and host factors are typically more difficult to control.

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

1. Contact transmission is the most important and frequent mode of transmission of health-care associated infections (HAIs). It can be divided into direct and indirect contact transmission.

• Direct contact transmission involves direct body surface-to-body surface contact resulting in physical transfer of microorganisms from an infected or colonized animal. For example, two dogs in a waiting room that come into direct contact when they sniff each other may transmit pathogens present in their noses or perineal areas; direct contact of a veterinarian's hands with a wound on an animal may result in transmission of opportunistic pathogens from the normal microflora of the person's hands, or infectious organisms present in the animal's wound, to the patient or the veterinarian, respectively.

• Indirect contact transmission is the result of physical transfer of microorganisms from the original animal (or human) source to a new host, without direct contact between the two. This typically involves body surface contact with an inanimate object, environmental surface or the integument of another animal or person that has been transiently contaminated by the original animal (or human) source. For example, handling one animal and then petting another animal without washing one's hands constitutes indirect contact between the two animals.

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

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

4. Vector-borne transmission occurs when vectors such as mosquitoes, flies, ticks, fleas, rats, and other vermin transmit microorganisms. Some act as simple mechanical vectors, comparable to indirect contact transmission, whereas others acquire and transmit microorganisms by biting.

It is important to have control measures in place to reduce or eliminate the presence of such vectors in veterinary clinics.

THE INFECTION CONTROL PROGRAM

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

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

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

• Help facilitate development of a written infection control manual

• Direct and document training of new staff (particularly lay staff)

• Perform formal or informal quality control evaluation of infection control practice compliance (e.g. observing cleaning and disinfection practices, hand hygiene)

• Perform ongoing surveillance and be the person designated to receive information about and record incidents of suspected hospital-associated infections.

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

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

In Veterinary Medical Centre, Gatton, ICPs are Janelle Davies (in training) and Viktoria Peter (in training) who are in the process of completing the necessary accredited training through an external provider. They are your first points of contacts.

For Dayboro Clinic, Heidi St John is your first point of contact.

2. SURVEILLANCE

Surveillance is a key component of any infection control program. Effective infection control is impossible without surveillance and should be performed by the ICP. It involves analysis of data that are already available (e.g. bacterial culture and susceptibility results, results of other kinds of infectious disease testing) to determine elements such as endemic disease rates, antimicrobial susceptibility patterns and trends, and changes in disease patterns. An example of passive surveillance would be monitoring the surgical site infection (SSI) rate following all surgical procedures and specific surgical procedures (e.g. spays, neuters). Monitoring of bacterial culture and susceptibility testing can provide information regarding possible outbreaks of hospital associated infections (HAIs), as well as information to guide empirical antimicrobial therapy. Routine recording of animals with specific syndromes such as vomiting, diarrhea, coughing or sneezing is another simple means of providing information that can help in the prevention and early detection of outbreaks, and can help to identify index cases should a hospital outbreak occur.

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

The keys to passive surveillance are to centralize the available data, and to have a designated infection control practitioner (ICP) who is responsible for compiling and evaluating this data on a regular basis. Simply collecting the data or even entering it in a spreadsheet is of no value unless someone looks at it. This is particularly important in large clinics or hospitals where multiple veterinarians may have patients with similar infections but do not communicate this to others, and therefore the start of an outbreak can be missed. If an outbreak is identified, then a plan can be formulated and implemented in order to stop the spread of disease. This plan may or may not include additional active surveillance to identify additional cases.

Current infection surveillance activities at the Veterinary Medical Centre, Gatton are coordinated by and include the following:

Passive Surveillance

Passive surveillance will be performed routinely via monitoring clinical samples sent from the Veterinary Teaching Hospital to the diagnostic laboratory, and providing monthly reports of zoonotic and antimicrobial resistant organisms. Clinical data associated with the cases are available for epidemiological analysis if necessary.

Active Surveillance

The presence of MDR *E.coli, Salmonella,* and MRSA in dogs and horses on admission to hospital will be determined by taking rectal and nasal samples when the animal is presented to the clinic. This will also ascertain how pathogens are introduced into the hospital. Initially, 200 animals will be sampled, which will enable detection of prevalence of 2%, 2-5% and 3-15%, respectively of these organisms. The sampling regime will be adjusted as required after baseline data is obtained. All swabs will be taken from dogs and horses with the owner's consent and providing the procedure does not compromise the health of the animal or hospital personnel.

- Obtain informed written consent from owner;

- Obtain rectal and nasal swab from the animal (labeled date, species, animal ID, source);
- Send samples to diagnostic laboratory with appropriate request slip.

Environmental surveillance: longitudinal surveillance

Environmental surveillance will initially occur over a 12-month period, commencing prior to the opening of the veterinary clinic to provide baseline data.

Baseline data: Environmental samples (n=400) have been taken from high risk areas such as drains, cages, floors and contact surfaces (doors, phones, shovels, railings, tables) or where it is important that the environment is free of pathogens e.g. surgeries and intensive care units.

- For the first 3 months after opening: 10-15 samples are to be taken weekly;
- For 3 – 6 months after opening: 10-15 samples to be taken fortnightly;
- After 6 months: 10-15 samples to be taken monthly.

This protocol will be modified depending on the actual prevalence of organisms. Environmental samples will be collected by swabs or electrostatic wipes. These samples will then be placed in a pre-enrichment broth. Total bacterial counts will determine the environmental load and *Salmonella,* MRSA and MDR *E.coli* will be identified and reported to Veterinary Teaching Hospital management and other stakeholders.

Evaluation of infection control strategies

Clinical staff will answer a questionnaire, to assess compliance and barriers to infection control, after 6 months, which will be followed by focus group interviews (chief nurse, interns, students). This information combined with the results of incidence of infections at admission, clinical infections, and environmental surveillance will evaluate biosecurity and infection control protocols.

Re-assessment and modification of infection control practices

If breakdowns in infection control are noted, further protocols for surveillance or infection control may need to be implemented. This is to mitigate the risk of infection and to maximise compliance with infection control procedures. Need for intervention or procedure modification can then be evaluated. If no interventions are necessary the objective will be to establish longer term surveillance and biosecurity protocols.

Swabbing of environmental and patient samples

Purpose

To ensure VTH staff are aware of and adhere to the necessary processes in place to ensure that clinical spaces are both monitored and maintained in a pathologically clean environment.

Procedure

1. In normal conditions 8 environmental sample swabs are to be taken monthly, early in the last week of the month.
2. Sampling areas are rotated as per excel spread sheet on (Y:\SVS\Operations\VTH-admin\Biosecurity\Environmental Surveillance\Environmental Surveillance Spreadsheet) and submitted to VLS.
    a. **4 x samples from SAH** and **4 x samples from ESH**
3. Environmental samples are to be submitted once monthly as a single submission to ensure a single charge to UQ VETS. Results will be submitted to the clinic as soon as possible, processing with C&S may take 2-4 days (or longer if fastidious).
4. For VLS test procedure and charging please refer Y:\SVS\Operations\VTH-Admin\SOP's\Internal Processes\Biosecurity, Infection Control, Hygiene & Cleaning\Biosecurity Plans\FORM VLS-0006 Environ sampling quote VMC.docx
5. Reports from the lab are to be saved in Y:\SVS\Operations\VTH-Admin\Biosecurity\Environmental Surveillance\VLS Lab reports
6. Sample site and results are to be logged in the Environmental Surveillance Spreadsheet.
7. If high risk situations arise within the hospital increased samples are to be taken ad hoc. E.g. sampling high risk areas post discharge of a patient known to have a MDR infection.
8. Any positive results from samples need to be communicated UQ VETS Lead Nurse *Gary Fitzgerald* so that appropriate action can be taken and recorded. Following this a repeat swab of the sample area must be taken at least a day after cleaning to be sure the bacteria has been fully eradicated.
9. Any post –operative infections identified should be identified and recorded in the database. The clinician in charge and UQ VETS Lead Nurse should be notified to identify any possible nosocomial links to allow appropriate action to be taken. E.g. antibiogram comparisons and/or further genetic testing to identify epidemiological links.

Staff responsible for environmental sample collection, submission, attaining and disseminating results:-

SAH: Janelle Davies
ESH: Gaby Doxey

VLS Test Protocols:

Environmental Screening: Swabs, Swipes

Environmental sample

- Transport media swab
- Swifter – to be placed in 100 ml of phosphate buffered saline (PBS) (cost swifter $6.90/20)
- Samples to be collected:

- Swabs/swifters incubated in PBS overnight.
- coliform counts and total cell counts performed from swifters only, not swabs
- MRSA
    - 1ml PBS into 9 mL enrichment broth (10 g/L Tryptone T, 75 g/L NaCl, 10 g/L mannitol, 2.5 g/L yeast extract) incubate overnight, 10 µL into Mannitol Salt agar with 2 µg/mL oxacillin or similar incubate overnight

        - Confirmatory tests (if requested) (Gram stain, VP, catalase, coagulase, PCR, not offered unless you are wanting to check clonality with previously isolated bacteria)
        - Disc diffusion (if requested)
        - *confirmatory ID and Sensitivities incur further charges*
- *Suspect MDR Enterobacteriaceae,*
    - 1ml PBS into 9 ml enrichment broth (TSB/Amp) incubate overnight, 10 µL onto MCA/Amp incubate overnight
        - Confirmatory tests as required
        - Disc diffusion (if requested)
        - *confirmatory ID and Sensitivities incur further charges*

-Pseudomonas species

- 20 µL of on to Pseudomonas cetrimide agar and incubate overnight
    - Confirmatory tests as required
    - Disc diffusion (if requested)
    - confirmatory ID and Sensitivities incur further charges

- *Salmonella Equine clinic only, unless otherwise requested*
    - 1 ml PBS into 9 mls enrichment broth (mannitol selenite) incubate overnight, 10 µL onto XLD, BGA incubate overnight
        - Confirmatory tests as required
        - Disc diffusion (if requested)
        - *confirmatory ID and Sensitivities incur further charges*

Associated Documents

The results from swabbing and actions taken to any positive results can be found at:
Y:\SVS\Operations\VTH-admin\Biosecurity\EnvironmentalSurveillance\EnvironmentalSurveillance Spreadsheet
Y:\SVS\Operations\VTH-Admin\SOP's\Internal Processes\Biosecurity, Infection Control, Hygiene & Cleaning\Biosecurity Plans\FORM VLS-0006 Environ sampling quote VMC.docxY:\SVS\Operations\VTH-Admin\SOP's updated\Internal Processes\Biosecurity & Infection Control\Biosecurity Plans.
Y:\SVS\Operations\VTH-Admin\SOP's updated\Internal Processes\Biosecurity & Infection Control\Infection Control

Refer to Y:\SVS\Operations\VTH-Admin\SOP's

3. ROUTINE PRACTICES

http://www.ava.com.au/sites/default/files/AVA_website/pdfs/Resource%202%20-%20Hand%20hygiene.pdf

Key points:

- Hand hygiene is the single most important way to prevent infections in the healthcare setting
- Bar soaps are not acceptable in veterinary practice settings
- It is recommended that jewellery not be worn on the hands during clinical work as it is difficult to wash hands adequately
- Finger nails: Natural nails more than 3-4 mm long are difficult to clean, can pierce gloves and harbour more microorganisms than short nails. Artificial nails or nail enhancements (including nail polish) should not be worn by anyone involved directly in patient care.
- Intact skin is the first line of defense against bacteria.
- No human food is to be kept in the clinical areas (and this includes nurses station) except for designated kitchen or dining areas. No eating or drinking in clinic areas except for designated areas.

Routine Practices are a way of thinking and of acting that forms the foundation for limiting the transmission of microorganisms in all health care settings.

Routine practices include:

- Hand hygiene
- Risk reduction strategies through use of personal protective equipment (PPE), cleaning and disinfection of the environment and equipment, laundry management, waste management, safe sharps handling, patient placement, and healthy workplace practices.
- Risk assessment related to animal clinical signs, including screening for syndromes that might indicate the presence of infectious disease (e.g. fever, coughing/sneezing, diarrhea, abnormal excretions/secretions), and use of risk assessment to guide control practices.
- Education of veterinary personnel and owners.

HAND HYGIENE

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

(a) 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. 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 in a disposable pump dispenser 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

Antibacterial soaps should be used throughout the hospital, particularly in high risk areas such as ICU. There are multiple hand washing stations situated throughout the Veterinary Medical Centre. These stations contain Microshield 4 (Chlorhexidine Gluconate 4%) in disposable pump dispensers. There are also alcoholic hand sanitiser bottles positioned around the clinic for all personnel to use intermittently throughout the day. Bar soaps are not acceptable in veterinary practice settings because of the potential for transmission of pathogens from one person to another.

Wash hands before and before and after every patient and after contact with any body fluids, potentially infectious or contaminated material; before and after contact with items in the patient's environment; before performing invasive procedures; before eating food and after personal body functions, such as using the toilet or blowing one's nose.

Recommended Technique for Hand Washing

- Remove all hand and arm jewelery.
- Wet hands with warm (not hot) water. Hot water is hard on the skin, and will lead to dryness and additional skin damage.
- Apply liquid or foam soap.
- 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 "Happy Birthday".
- Using a rubbing motion, thoroughly rinse soap from hands under warm running water. Residual soap can lead to dryness and cracking of skin.
- Dry hands thoroughly by blotting hands gently with a paper towel. Rubbing vigorously with paper towels can damage the skin.
- Turn off taps with paper towel to avoid recontamination of your hands.

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

It is recommended that jewellery not be worn on the hands during clinical work as it is difficult to wash hands adequately; rings can be kept on a neck chain in these circumstances); short-

sleeved clothing or sleeves to the elbow at maximum is recommended to facilitate adequate handwashing.

(B) USE OF ALCOHOL-BASED HAND SANITIZERS

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

Alcohol-based hand sanitizers should contain 70-90% alcohol. Use of products containing emollients helps to reduce skin damage which can otherwise occur with frequent use of hand sanitizers. Products containing alcohol and chlorhexidine are also available. Chlorhexidine provides some residual antimicrobial action on the hands after use, but it is unclear whether or not these combinations provide any true benefit in clinical settings. They may be more useful as alternatives to traditional surgical scrubbing techniques (see Surgery).

Alcohol-based hand sanitizers are not effective against certain pathogens, including bacterial spores (e.g. clostridial spores) and Cryptosporidium spp. Nonetheless, alcohol-based hand sanitizers may be useful even if alcohol-resistant pathogens like Clostridium difficile are present. The improved hand hygiene compliance seen with alcohol-based hand sanitizers and their efficacy against other pathogens are important aspects of infection control Routine use of these products has not resulted in detectable increases in C. difficile infection rates in human hospitals. However, if hands are potentially contaminated by one of these organisms, hand washing with soap and running water should be performed if possible. Although even 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. Alcohol is also not as effective against non-enveloped viruses (e.g. canine parvovirus, feline panleukopenia virus) 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.

Recommended Technique for Alcohol Based Sanitizers

- Remove all hand and arm jewelery.
- Ensure hands are visibly clean (if soiled, follow hand washing steps).
- Apply between 1 to 2 full pumps or a 2-3 cm diameter pool of the product onto one palm.
- 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.
- Rub hands until product is dry. This will take a minimum of 15 to 20 seconds if sufficient product is used.
- Hands must be fully dry before touching the patient or patient's environment/equipment for the hand rub to be effective, and to eliminate the rare risk of flammability in the presence of an oxygen-enriched environment, as may occur in the presence of gas anesthetic machines.

FACTORS THAT INFLUENCE THE EFFECTIVENESS OF HAND HYGIENE

• Condition of the skin: Intact skin is easier to clean than skin that is chapped, cracked, cut, abraded or otherwise inflamed. Intact skin is the first line of defense against bacteria.

• Finger nails: Natural nails more than 3-4 mm long are difficult to clean, can pierce gloves and harbour more microorganisms than short nails. Artificial nails or nail enhancements (including nail polish) MUST not be worn by anyone involved directly in patient care, as they have been implicated in the transfer of microorganisms in human medicine.

• Jewelery: Jewelery is very hard to clean, and physically protects bacteria and viruses from the antiseptic action of alcohol-based hand sanitizers and the mechanical cleaning action of soap and running water. Rings 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.

SKIN CARE

Careful attention to skin care is an essential part of the hand hygiene program. Products used for hygiene should be "hand-friendly" – for example, alcohol-based hand sanitizers containing emollients are available, which can help reduce the drying effect of the alcohol. If skin integrity is an issue, the individual should consult his or her physician. Skin lotions can help maintain the health and integrity of the skin, but it is important to use a skin lotion that does not interfere with glove integrity. Petroleum-based lotion formulations can weaken latex gloves and increase permeability. Lotions that contain petroleum or other oil 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.

Intact skin is the first line of defense against bacteria.

4. PERSONAL PROTECTIVE EQUIPMENT (PPE)

http://www.ava.com.au/sites/default/files/AVA_website/pdfs/Resource%201b%20-%20How%20to%20protect%20yourself%20small%20animal%20practice.pdf

http://www.ava.com.au/sites/default/files/AVA_website/pdfs/Resource%203%20-%20Sequence%20for%20putting%20on%20PPE.pdf

An abstract from guidelines from AVA: "Street clothes should always be covered by protective outerwear, such as a lab coat, when working in the clinic."

Key points:

- Some form of PPE must be worn in all clinical situations, including any contact with animals and their environment.
- Protective outerwear, including scrubs and lab coats, MUST not be worn outside the clinic.
- Gloves must be worn if warranted in the judgement of the senior clinical staff when handling animals to minimize the potential exposure to agents with zoonoses hazards
- P2 masks should be worn if in warranted in the judgment of the senior clinical staff, eg if there is likely to be contact with body fluids or transmission of a zoonotic disease by aerosol (eg Bordetella).
- Special care should be taken in the case of personal risk factors such as possible immunosuppression in persons with conditions which may affect the immune system, or taking immunosuppressive drugs.
- Closed-toed footwear must be worn at all times to reduce the risk of injury from various sources.

Personal protective equipment (PPE) is an important routine infection control tool. PPE use is designed to reduce the risk of contamination of personal clothing, reduce exposure of skin and mucous membranes of veterinary personnel to pathogens, and reduce transmission of pathogens between patients by veterinary personnel. Some form of PPE must be worn in all clinical situations, including any contact with animals and their environment. These recommendations must always be tempered by professional judgment, while still bearing in mind the basic principles of infectious disease control, as every situation is unique in terms of the specific clinic, animal, personnel, procedures and suspected infectious disease.

Personal protective outerwear is used to protect veterinary personnel and to reduce the risk of pathogen transmission by clothing to patients, owners, veterinary personnel and the public. Protective outerwear should be worn whenever there may be contact with an animal or when working in the clinical environment (including cleaning).

Here are some of the basic personal practices to be considered by clinic personnel and students:

- Long hair should be tied back.
- Closed-toe shoes which are easily cleaned should be worn.
- Fingernails should be short to make washing and scrubbing effective therefore no fake or acrylic nails.

Special care should be taken in the case of personal risk factors such as possible immunosuppression in persons with conditions which may affect the immune system, or taking immunosuppressive drugs (cortisone, cyclosporine etc); wounds, cuts & scratches; chronic or acute intercurrent medical conditions (eg colds and influenza, asthma, eczema, respiratory disease, diabetes and others), pregnancy. Avoid touching the face with hands to minimise likelihood of germ transfer. Protective clothing such as scrubs tops, scrubs, gowns, overalls or lab coats should be worn. Protective clothing must be changed if grossly contaminated or if an animal with a known infectious disease is contacted. Fresh protective clothing must be worn every day. Used protective clothing should be put in a suitable bag separate from clean items until it is laundered. Students to provide their own protective clothing which should be kept separately from other clothing and equipment after wearing (eg in a garbage bag) and laundered in hot water after each use.

Gloves and P2 masks should be worn if in warranted in the judgment of the senior clinical staff, eg if there is likely to be contact with body fluids or transmission of a zoonotic disease by aerosol (eg Bordetella). Protective eye wear should be born if there is a possibility of contamination of the eyes with organic material or a pathogen (eg during orthopaedic or dental procedures). Gloves do not preclude the necessity of regular handwashing between each animal. Special protective equipment should be used if there is a higher risk, eg heavy gloves for handling bats given a Biosafety approval is in place from UQ Institute for Biosafety (for Australian Bat Lyssavirus, Hendra Virus), other wildlife or fractious cats (cat scratch).

No human food is to be kept in the clinical areas except in the designated areas. Eating or drinking should never occur in patient care, samples or instrument processing areas or where hazardous chemicals are handled. Separate refrigerators should be used for human food, animal food, and biologics. Dishes for human use should be cleaned and stored away from animal care areas.

An abstract from guidelines from AVA: "*Protective outerwear, including scrubs and lab coats, should not be worn outside the clinic".*

Personnel should change into clinic clothes at the beginning of their shift and back into street clothes at the end of their shift.

LAB COATS/ CONSULTING JACKETS

Front-closing lab coats are meant to protect clothing from contamination, but generally they are not fluid resistant, so they should not be used in situations where splashing or soaking with potentially infectious liquids is anticipated. These garments should be changed promptly whenever they become visibly soiled or contaminated with body fluids, and at the end of each day. Lab coats worn in the clinic should not be worn outside of the work environment. Lab coats worn when handling patients with potentially infectious diseases should be laundered after each use, because it is almost impossible to remove, store/hang and reuse a contaminated lab coat without contaminating hands, clothing or the environment.

SCRUBS

Scrubs are often worn in veterinary clinics as a form of basic personal protective equipment. They have the advantage of being durable and easy to clean, and their use prevents

contamination and soiling of the street clothes that personnel wear outside the clinic. Clinic scrubs should not be worn outside the clinic. They should not be taken home by personnel to be washed, rather they should be washed on- site, with other clinic laundry. Scrubs should be washed at the end of each day and whenever they become visibly soiled. If scrubs are brought home, they should be kept in a plastic bag until being placed in the washing machine, and washed separately from other laundry in hot water and detergent.

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

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

Protective outerwear, including scrubs, should not be worn outside the clinic.

NON-STERILE GOWNS

Gowns provide more coverage for barrier protection than lab coats, and are typically used for handling animals with suspected or confirmed infectious diseases, that are housed in isolation. Permeable gowns can be used for general care of patients in isolation. Impermeable (i.e. waterproof) gowns should be used to provide greater protection when splashes or large quantities of body fluids are present or anticipated. Disposable gowns should not be reused, and reusable fabric gowns should be laundered after each use, because hanging/storing and reusing contaminated gowns inevitably leads to contamination of hands, clothing or the environment. Gloves should be worn whenever gowns are worn. Gowns (and gloves) should be removed and placed in the trash or laundry bin before leaving the animal's environment, and hands should be washed immediately afterwards.

Personnel should learn to remove gowns properly, in such a way as to avoid contaminating themselves and the environment. All gowns should be used only once, then discarded or laundered.

GLOVES

http://www.ava.com.au/sites/default/files/AVA_website/pdfs/Resource%204%20-%20Gloves.pdf

Gloves reduce the risk of pathogen transmission by providing barrier protection. They should be worn when contact with blood, body fluids, secretions, excretions and mucous membranes is possible. Gloves should also be worn when cleaning cages and environmental surfaces, as well as when doing laundry if gross contamination of items is present.

Gloves should be removed promptly after use, avoiding contact between skin and the outer glove surface. Gloved hands should not be used to touch surfaces that will be touched by people with non-gloved hands. Care should be taken to avoid contamination of personal item such as telephones, pens and pagers. Hands should be washed or an alcohol-based hand sanitizer should be used immediately after glove removal. It is a common misconception that using disposable gloves negates the need for hand hygiene. Gloves do not provide complete protection against hand contamination, therefore hand hygiene immediately after removing gloves is essential.

Disposable gloves should not be washed and reused. Gloves are NOT a substitute for proper hand hygiene.

Change gloves and perform hand hygiene when:

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

Latex gloves are commonly used, but if latex allergies are a concern, acceptable alternatives include nitrile or vinyl gloves. Latex gloves will decompose and lose come in a variety of materials. The choice of glove material depends on their integrity when exposed to many chemicals. If exposure to chemicals such as disinfectants is expected (e.g. when cleaning and disinfecting cages), disposable nitrile gloves or heavier, reusable rubber gloves (e.g. common dishwashing gloves) can be used. Reusable gloves must also be disinfected at the end of each task.

FACE PROTECTION

Face protection prevents exposure of the mucous membranes of the eyes, nose and mouth to infectious materials. Face protection typically includes a nose-and-mouth mask (e.g. surgical mask) and goggles, or a full face shield, which should be used whenever exposure to splashes or sprays is likely to occur, including dental procedures, nebulization, and wound lavage.

RESPIRATORY PROTECTION

Surgical masks do not provide respiratory protection and are not a replacement for respirators.

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

FOOTWEAR

Closed toed footwear must be worn at all times to reduce the risk of injury from dropped equipment (e.g. scalpels, needles), scratches from being stepped on by dogs, and to protect the feet from contact with potentially infectious substances (e.g. faeces, discharges and other body fluids).

Designated footwear or disposable shoe covers are required in areas where infectious materials are expected to be present on the floor, in order to prevent their spread to other areas. This is

particularly important in veterinary clinics because patients, and sometimes the personnel working with them, often have very close contact with the floor, unlike human hospitals. Designated footwear or disposable shoe covers may be required for patients with infectious diseases that are kept on the floor (e.g. in a large dog run) or that may contaminate the floor around their kennel (e.g. an animal with severe diarrhea). Such footwear must be removed as the person leaves the contaminated area, and should be immediately disposed of in the garbage (if disposable), or left at the entrance of the contaminated area on the “dirty” side.

In veterinary clinics, it is important to prevent the spread of infectious materials present on the floor, as patients and personnel often have very close contact with the floor.

5. CLEANING AND DISINFECTION

Gloves should be worn when cleaning and disinfecting, and hands should be washed after finishing any cleaning activity.

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

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

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

CLEANING

Cleaning entails the removal of all forms of organic matter (e.g. feces, urine, blood, food, dirt etc.) from a surface. Ensure all areas are well ventilated during cleaning. Cleaning must always be done before a disinfectant is used. After cleaning, allow all surfaces to dry completely.

Avoid generating airborne dust that may contain pathogens by:

- Using a vacuum cleaner equipped with a HEPA filter. The filter helps to prevent aerosolisation of pathogens such as ringworm. For this reason, vacuums without HEPA filters should not be used for cleaning in patient-contact areas.
- Lightly spraying surfaces with water prior to mopping or sweeping using an electrostatic wipe (e.g. SwifferTM cloth) using a wet mop.
- Exposure to aerosols generated by brushes during cleaning can be minimized by taking certain precautions, such as wearing a P2 mask and containing spatter if the brush or surface is damp. A surgical nose-and-mouth mask will provide some protection against droplet spatter, but not against finer particles and dry dust that can become suspended in the air.
- Removing sticky, wet or dried-on organic material from surfaces: This kind of debris should be removed using a detergent or soap and a brush or cloth, as necessary. During cleaning, it is the mechanical action and surfactant properties of the soap that are important, not necessarily its antimicrobial activity.

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

DISINFECTION

Disinfection is more effective if preceded by thorough mechanical cleaning. Ensure the area is well ventilated before using disinfectants. Gloves should be worn when handling disinfectants, but latex gloves will decompose and lose their integrity when exposed to many chemicals. For small jobs, disposable nitrile gloves should be used instead. For large jobs, heavier rubber gloves (e.g. common dishwashing gloves) can be used, but reusable gloves of this type must also be disinfected at the end of each task.

Always refer to the product label with respect to dilution rates and required contact time. For general cleaning purposes, UQ Veterinary Medical Centre utilises Value Plus Stable and Kennel Disinfectant (10mls:500mls water). This solution is used only a general cleaner, and should not be used as a high grade disinfectant. For infectious diseases (e.g. Canine Cough, Cat Flu) UQ SAH & Teaching Hospital uses either F10, Virkon S* or or Oxivir (Broad Spectrum virucidal/bactericidal/fungicidal disinfectant).

*Refer to SOP on Disinfectants:
Y:\SVS\Operations\VTH-Admin\SOP's\Internal Processes\Biosecurity, Infection Control, Hygiene & Cleaning

Reception areas

Reception areas are the public face of the clinic and create the all-important first impression for clients. Reception areas should be noticeably clean and fresh.

Specifically:

All surfaces damp-dusted daily using an appropriate disinfectant. Surfaces which are touched by people (e.g. door handles and door plates, front and top of reception counter) should be regularly cleaned with alcohol spray and dried.

Floors should be vacuumed at least daily and more often if any noticeable amount of hair accumulates during the day. Hair should be vacuumed from the floor using a vacuum cleaner equipped with a hepa-filter to minimize spread of potential pathogens such as ringworm spores into the environment. Vacuuming should be performed before floor washing.

Floors must be washed regularly with a suitable detergent (to remove grease and oils) and a germicidal disinfectant (one which kills parvovirus). During the day, any organic matter on the floor (urine, saliva, faeces, vomitus) or any liquid (eg solution spills) must be cleaned promptly. Clean-up areas should be either dried immediately with paper or cloth towel, or any slip hazard identified and taped off and marked as a hazard until the floor is dry.

Any display shelving must be damp dusted using a suitable disinfectant at least weekly and preferable more often. Shelf contents must be similarly kept clean.

Toys should be of a material which can be disinfected. Soft toys are unsuitable for reception areas where they may be handled by many children or their carers. Toys should be disinfected as least weekly, eg by immersion in a bleach solution and natural drying, more often if indicated. Toy boxes should be cleaned with bleach or alcohol at least weekly.

A designated waiting area should be available for any animal which might have an infectious disease. This area should be away from other patients, preferably in another room or outside. For example, animals with acute cough (Bordetella), diarrhea (Canine parvovirus), or skin lesions (ringworm) should be segregated from other patients while waiting.

Consulting rooms

Consulting rooms are the public face of clinical practice in the hospital. The environment in the consulting room is keenly observed by clients and procedures must not only minimise the risk of spread of infection but be seen to do so. Infection control is as for reception areas and additionally:

Examination table surfaces must be sprayed with F10, Virkon or Oxivir and dried with paper towel after every animal.

Any area which becomes contaminated during a consultation (eg sink bench after minor procedures such as ear cleaning, corneal staining, venepuncture) must be sprayed with alcohol and wiped between every animal examined and treated.

Sinks and benches in consulting rooms must be cleaned thoroughly with a standard cleaner which will lift grease, oil, and water-soluble contaminants (for example Jif or Ajax), finished with alcohol spray and wiped dry at least daily.

All surfaces in consulting rooms including table legs, shelves, equipment trolleys etc must be cleaned at least weekly, including cleaning with alcohol spray or other suitable disinfectant. Everything on shelves and trolleys must be moved for effective cleaning. Equipment, models, books, leaflets, etc should be cleaned or damp-wiped as appropriate.

Floors must be vacuumed and washed daily and spills and contamination removed promptly as for floor care in the reception areas. It is useful to have easy access to a small vacuum cleaner in the consulting room.

Disinfectants should be kept in the consulting room. Paper towels rather than cloth towels should be used and appropriate yellow biohazard bags for disposal of organic waste located in the consulting room. If cloth towels are used they should be used only once and disposed of immediately after use into an appropriate laundry container.

Hospital, preparation and theatre areas

Hands must be washed after any animal, its bedding or equipment is touched. Cages must be changed regularly at least daily, and soiled bedding or equipment changed promptly. As for consulting rooms, all sharps to be disposed of immediately in sharps containers, soiled dressings or organic material to be disposed of in yellow biohazard bags. Regular cleaning of all surfaces, shelves, trolleys, etc as for consulting rooms.

Isolation wards

Scrupulous cleaning and hygiene as for other areas plus:

Change of shoes OR shoe covers on entry; change of shoes OR remove booties on exit;

Wash hands on entry and exit;

Fresh gown on entry, to be removed on exit. (If consideration is given to leaving the same gown ‘hung up’ in the isolation ward, extreme care should be taken not to contaminate clothing with the outside of the gown).

Gloves should be worn if touching an animal or its bedding or equipment.

Consideration given to masks (P2 if necessary) and protective eyewear.

Isolation wards to have their own equipment – thermometers, stethoscopes etc, must be disinfected after each use and if brought outside isolation ward.

6. SINGLE-USE VS REUSABLE EQUIPMENT

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

Multi-use equipment must be properly cleaned and disinfected between each patient.

In veterinary medicine, exceptions to the level of processing required are typically made for some pieces of semi- critical equipment that come in contact with tissues or mucous membranes which are normally considered non- sterile, such as those of the upper respiratory or gastrointestinal tracts. If a transmissible infectious disease is not suspected in the patient, and the subsequent patient is not significantly immunocompromised, thorough cleaning and low level disinfection is likely adequate in these cases. However, if an infectious disease is suspected or the subsequent patient is immunocompromised, then cleaning and high level disinfection or sterilization are recommended in order to prevent disease transmission.

Food and water bowls of patients with infectious diseases should be cleaned and disinfected separately, but careful selection of the disinfectant used is required because only some disinfectants are approved for use on surfaces that come in contact with food. Otherwise disposable dishes can be considered for these animals. Cleaning alone (with regular dish soap) is adequate for food and water bowls from other patients. Toys, litter boxes, and other miscellaneous items should be cleaned and disinfected between patients, or discarded if they are not amenable to proper cleaning and disinfection. Gloves should be worn when handling items from patients carrying zoonotic pathogens or any items that are visibly soiled. Litter boxes should be cleaned out at least daily, and completely emptied and disinfected between patients. Ideally, litter boxes should not be handled by pregnant women, however if daily cleaning and disinfection are performed properly, the risks are minimized.

COLD STERILIZATION

“Cold sterilization” is used to sterilize items through immersion in a sterilizing solution. Because of the toxicity of some cold sterile solutions, the time required to achieve sterilization using these chemicals, and the availability of autoclaves for sterilization, there is minimal indication for the use of cold sterilization. Its main indication is for sterilization of items that cannot tolerate steam sterilization, such as endoscopes.

Although cold sterilization can be an effective means of sterilizing instruments, misuse can result in ineffective sterilization. Potential problems include the use of inappropriate solutions, improper preparation of solutions (i.e. inadequate concentration), inadequate contact time, inadequate replacement/refreshment of solution, or inadequate removal of organic debris from equipment prior to immersion in solution. Commonly used disinfectants such as alcohol, iodophors, phenolics and most quaternary ammonium compounds are not effective sterilants

and therefore are not acceptable for use on items intended to be used in surgical or other invasive procedures.

Prolonged contact time (e.g. 10 hours) is required for sterilization using these solutions. Therefore, cold sterilization is not a means for rapid sterilization of surgical instruments that have been inadvertently contaminated during surgery or for surgical instruments that will be used frequently on different patients throughout the day. In some veterinary clinics, disinfectant solutions of other kinds in which a set of instruments is routinely kept are frequently referred to as “cold sterile.” Such misuse of this term should be avoided, as instruments kept in disinfectant solutions other than glutaraldehyde or high-level sterilants should not be used for surgical or other invasive procedures.

Instruments must be cleaned to remove all visible organic debris (including blood) before placing them in a clean, fresh cold sterilant solution in order for the procedure to be effective. Most chemical sterilants come in solutions consisting of two parts that, when combined, form what is referred to as an "activated" solution. Refer to the product’s label for the shelf life of the activated solution. Cold sterilant must be rinsed off all instruments using sterile saline or water before they are used, as some of these compounds (particularly glutaraldehyde) can be irritating to tissues. As with all other chemicals used in a veterinary clinic, Material Safety Data Sheets (MSDS) for these products must be readily available to all personnel who work with them and around them.

MAINTENANCE OF ENDOSCOPES

Proper cleaning and maintenance of endoscopes are important to prolonging the useful life of the instrument, but cleaning and disinfection are also important from an infectious disease control aspect. Endoscopes are semi-critical equipment, and as such require high level disinfection when used in humans. In veterinary medicine, high level disinfection is required prior to use in relatively sterile areas (e.g. urinary tract), but thorough low level disinfections is considered adequate for use in non-sterile areas (e.g. gastrointestinal tract, upper respiratory tract) if a transmissible infectious disease was not suspected in the previous patient and the subsequent patient is not significantly immunocompromised. Manufacturers typically provide detailed reprocessing (cleaning and disinfection) instructions for their instruments, which should be readily available as a reference for staff members responsible for the care of endoscopes. If the endoscope was purchased second hand and the reprocessing instructions were not provided, it is important to contact the manufacturer to obtain a copy. Some general guidelines regarding endoscope maintenance include:

• Endoscopes must be meticulously cleaned immediately after every use. Endoscopes typically have several moving or detachable parts and small channels in which moisture, debris and discharge can become trapped. Cleaning must be performed as soon as possible in order to prevent debris from drying onto surfaces, as this can make the debris considerably harder to remove. Prior cleaning is crucial to effective disinfection.

• All instrument and suction channels must be thoroughly cleaned after each use, even if the channels were not used during the procedure. Failure to clean these channels is a common error which can result in accumulation of debris, bacteria and biofilms within the instrument. Not only does this pose risk of disease transmission to subsequent patients, but it can also confound sample collection and culture.

• Rinsing and drying of the endoscope are also critical to proper maintenance. Failure to rinse off detergents or disinfectants can lead to significant irritation of the tissues of the next patient.

• Chemical sterilants (e.g. ortho-phthaladehyde) are typically used for high-level disinfection or sterilization of endoscopes, as most cannot be steam-sterilized (autoclaved). Consult the manufacturer's instructions regarding what methods can be safely used for any particular endoscope. If a chemical sterilant is used, a timer should be used to measure the exact contact time – too short a time may result in an inadequate microbial killing, while too long a time may result in damage to the instrument.

MAINTENANCE OF CLIPPERS

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

Clippers should be thoroughly cleaned and disinfected at the end of each use, before they are used for another patient. First, a stiff brush should be used to remove visible dirt and hair from the blade. The clipper blades can be further cleaned and disinfected using an appropriate blade wash product, before being soaked with alcoholic-chlorhexidine for two minutes. Then they are oiled and returned to patient use.

Contaminated blades are classified as those that have come into contact with infectious materials such as infected skin or faecal matter. Contaminated clipper blades should be first cleaned with an F10 soaked gauze to remove gross debris, before being sprayed with F10 and wiped clean of contaminates. They are then soaked in Surgistain solution for 15minutes before being rinsed with distilled water, dried thoroughly and oiled with an appropriate clipper oil.

The body of the clippers should be wiped with a standard disinfectant solution (F10), paying particular attention to the small crevices of the device and allowing for adequate contact time with the disinfectant. Refer to the clipper's instruction manual to determine what degree of contact with liquid the clippers can safely withstand.

LAUNDRY

Although soiled linens are a potential source of microorganisms, with appropriate hygienic handling, storage and processing of clean and soiled linens, the risk of disease transmission from these items can be reduced to an almost negligible level.

Linens and special clothing used in veterinary clinics (e.g. cage blankets, towels, surgical drapes, surgical gowns, scrubs, lab coats) can be an important means of transporting pathogens from one area to another within the clinic, and to areas outside the clinic.

While the Clinic clothing which are grossly contaminated must be laundered on site, staff and students should place their clothing in a separate bag to be taken home and launder on hot cycle.

Personnel should change into clinic clothes at the beginning of their shift and back into street

clothes at the end of their shift.

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

COLLECTION AND HANDLING

Except for linens potentially contaminated with infectious agents (see below), all used linens can be handled in the same way. Heavily soiled linens should be rolled or folded to contain the heaviest contamination in the centre of the bundle, without contaminating personal clothing or the environment. Large amounts of solid debris, faeces or blood clots should be removed from linen with a gloved hand and disposable tissue or paper towel, which are then immediately placed in the garbage. Excrement should not be removed by spraying with water or shaking as this may result in contamination of the surrounding area and personal clothing.

BAGGING AND CONTAINMENT

• Linens should be handled with a minimum of agitation and shaking.

• Always place soiled linens directly in a hamper or bag designated for dirty laundry.

• Never place soiled linens on the floor.

• Laundry bags should be tied securely and not over-filled.

• Carts and hampers should be cleaned after each use.

• Laundry bags should be washed after each use. They can be washed in the same cycle as the linens they contain. Alternatively disposable bags can be used for holding laundry and replaced regularly.

TRANSPORT

Linen transported by cart should be moved in such a way that the risk of cross-contamination is minimized (e.g. avoid moving the cart from potentially contaminated areas (runs/kennel area) to cleaner areas (prep room, surgery).

Clean linen should be transported and stored in a manner that prevents contamination. If laundry carts are used, separate carts should be used for clean and dirty linens.

Linens contaminated with gross organic material must be pre-cleaned by hand to remove such material prior to laundering. It is not possible to adequately clean laundry by machine when gross organic material is present, and laundering such items can lead to contamination of other laundered items.

WASHING AND DRYING

• Use of normal machine washing with a commercial laundry detergent and machine drying are sufficient to greatly reduce the numbers of most significant infectious pathogens from most soiled linens.

• If laundry is washed in cold water, an appropriate cold-water detergent must be used according to label directions.

• It should not be assumed that hot water washing will disinfect or sterilize items. High temperature (> 71.1^{o}C) washing can significantly reduce bacterial numbers, but standard household washing machines do not typically reach this temperature, even if the hot water setting is used.

• The heat and drying effects of tumble drying are a critical step in the laundering process, and account for a large proportion of the decrease in bacterial counts achieved. Therefore, laundry should not be considered clean until it has also been dried completely, ideally using the highest heat possible.

Line-drying linens outdoors may have the advantage of also exposing the surface of the fabrics to ultraviolet (UV) light, if they are hung to dry in the sun. However, it would be difficult to expose all surfaces to sunlight, and thick fabrics, items made of multiple fabric layers and those containing seams may protect bacteria from UV exposure. Also, the antimicrobial action of the high heat of tumble drying is lost if linens are line-dried, therefore tumble drying is recommended, especially for any materials that may have been contaminated with a transmissible infectious pathogen.

LAUNDRY FROM INFECTIOUS CASES

• Laundry from potentially infectious cases should be treated separately from other laundry.

• Linens should be collected in a separate linen bag and washed and dried separately.

• For linens with gross contamination of a potentially infectious nature (e.g. faeces from a diarrheic animal, discharge from an infected wound, urine from an animal with a urinary tract infection), as much organic material as possible should be removed by hand (using gloves and disposable tissue or paper towel, as described above). The items should then be pre-soaked in bleach solution (9 parts water:1 part household bleach) for 10- 15 minutes prior to machine washing.

• Disposal through Pathological waste (including double bagging) can be considered.

• Bleach should also be added to the household detergent in the washing machine as per label instructions.

PROTECTION OF PERSONNEL

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

Laundry should not be considered clean until it has also been dried.

WASTE MANAGEMENT

Veterinary biomedical waste is a potential source of both zoonotic and non-zoonotic infectious pathogens. Therefore, it is important to handle all such waste appropriately. Biomedical waste

typically includes sharps, tissues (anatomic waste), highly contaminated (e.g. blood-soaked) materials, and dead animals.

Used sharps are considered biomedical waste and should be disposed of in approved, puncture-resistant sharps disposal containers to remove, store and dispose of used sharps such as needles, blades, razors and other items capable of causing punctures. Non-anatomical waste saturated or dripping with blood (e.g. blood-soaked lap sponges and gauze) are also best disposed of as biomedical waste. If there is likely to be splashes or sprays during this disposal process, appropriate personal protective equipment should be worn.

All other waste, such as general office waste and non-sharp medical equipment, may be disposed of in the regular waste stream, and requires no special treatment other than containment during disposal and removal. Waste should be contained in a leak-proof container or bag that can be discarded with the waste (e.g. a plastic garbage bag).

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

Precautions should be taken to minimize contamination of the clinic environment and the risks to people and animals from potentially infectious waste.

Companion animal infectious diseases of Concern[1]

Bordetella spp infection
Canine parainfluenza virus
Canine influenza
Canine infectious hepatitis
Feline calicivirus
Feline herpesvirus
Feline panleukopaenia virus
Canine leptospirosis
Campylobacter spp infection
Salmonella spp infection
Cryptosporidium and Giardia
Psittacosis (Chlamydia psittici (avian))
Feline Leukaemia
Feline Immunodeficiency Virus
Cat scratch disease (Bartonella henselae & other bacteria)
Methcillin Resistant Staphlococcus Aureus infection

[1] This list not exhaustive.

Australian Bat Lyssavirus (not reported clinically in dogs & cats but develop antibodies after experimental exposure)
Canine distemper
Toxoplasma gondii
Giardia spp
Roundworms - Toxocara canis & Toxascaris spp
Hookworms – esp Ancyclostoma caninum
External parasites (fleas, ticks & mites)
Canine parvovirus

7. SURGERY

All surgical procedures cause breaks in the normal defensive barriers of the skin or mucous membranes. These breaks are therefore accompanied by an inherent risk of surgical site infection (SSI). Surgical site infections can occur sporadically, or as part of an outbreak and can have devastating outcomes in some situations. Good general infection control practices (e.g. hand hygiene, cleaning and disinfection) are important for prevention of SSIs. Specific measures pertaining to surgery include proper maintenance of surgical environment, use of appropriate personal protective equipment, hand hygiene, disinfection and sterilisation of anaesthetic equipment and surgical instruments, appropriate use of peri-operative antimicrobials, and surgical site care before, during and after the procedure.

Introduction

The use of sterile gloves does not render surgical hand preparation unnecessary. Sterile gloves contribute to preventing surgical site contamination and reduce the risk of blood-borne pathogen transmission from patients to the surgical team. However, 18% (range: 5–82%) of gloves have tiny punctures after surgery, and more than 80% of cases go unnoticed by the surgeon. After two hours of surgery, 35% of all gloves demonstrate puncture, thus allowing water and body fluids to penetrated the glove. A recent trial demonstrated that punctured gloves double the risk of SSIs.

SURGICAL ENVIRONMENT

A surgical theatre should only be used for surgical procedures. Having a well-designed and maintained surgical theatre or suite is very important. In order to keep the surgical environment as clean as possible, this area should be separated from personnel and animal traffic, and be easy to thoroughly clean and disinfect. A surgical theatre should not be used for non-surgical procedures between surgeries. Entrance to the area should be restricted at all times to minimize traffic in the room. The number of people in the surgical area has been identified as a risk factor for SSI in small animals, so only essential personnel should be allowed in the surgical theatre during any surgical procedure. All personnel participating in the procedure, including those performing surgical nursing duties, must be trained in aseptic technique and operating room procedures.

PERSONNEL CONSIDERATIONS

PERSONAL PROTECTIVE EQUIPMENT

All personnel in the surgical area should wear freshly laundered designated surgical scrubs, a surgery cap or hair bonnet, beard cover (if applicable) and a surgical mask (fitted over the mouth and nose) when surgery is underway, regardless of whether or not they are directly involved in the procedure itself. Designated surgical shoes or shoe covers must be worn by all personnel in the surgical area. Freshly laundered surgical scrubs worn in surgery should not be worn when handling or treating other patients and at a minimum should be covered with a lab coat when outside the surgery area (see Personal Protective Equipment under Routine Procedures). Personnel directly involved in the procedure should also wear a sterile gown and sterile gloves.

HAND HYGIENE AND SURGICAL SCRUB

Selection of products for surgical hand preparation

Agents used for surgical hand preparation should significantly reduce microorganisms on intact skin, contain a non-irritating antimicrobial preparation, have broad-spectrum activity, and be fast-

acting and persistent. The most commonly used products for surgical hand antisepsis are chlorhexidine or povidone-iodine-containing soaps. The most active agents (in order of decreasing activity) are chlorhexidine gluconate, iodophors, triclosan, and plain soap.

Application of chlorhexidine or povidone-iodine result in similar initial reductions of bacterial counts (70–80%), reductions that achieves 99% after repeated application. Rapid regrowth occurs after application of povidone-iodine, but not after use of chlorhexidine. Hexachlorophene and triclosan detergents show a lower immediate reduction, but a good residual effect. These agents are no longer commonly used in operating rooms because other products such as chlorhexidine or povidone-iodine provide similar efficacy at lower levels of toxicity, faster mode of action, or broader spectrum of activity. Despite both in vitro and in vivo studies demonstrating that it is less efficacious than chlorhexidine, povidone-iodine remains one of the widely-used products for surgical hand antisepsis, induces more allergic reactions, and does not show similar residual effects.

Surgical hand preparation is necessary to minimise the risk of resident or transient microorganisms contaminating the surgical wound.

Artificial nails, nail polish and jewellery on the hands and wrists must be removed before commencing the surgical scrubbing procedure.

A surgical hand scrub should be performed before putting on a sterile gown and sterile gloves. Various surgical scrub techniques have been described. Most commonly, a structured five minute surgical scrub with Chlorhexidine or Iodine generally is used, however Avagard hand scrub can be used by surgeons if a full sterile hand scrub has been performed prior that day.

SURGICAL SCRUB TECHNIQUE

Required time for the procedure

In 1939, Price suggested a 7-minute handwash with soap, water, and a brush, followed by 70% ethanol for 3 minutes after drying the hands with a towel.[63] In the second half of the 20th century, the recommended time for surgical hand preparation decreased from >10 minutes to 5 minutes.[509–512]Even today, 5-minute protocols are common.[197] A comparison of different countries showed almost as many protocols as listed countries.

Comparison of hand bacterial counts after 3-minute and 5-minute scrubs with seven different formulations showed that the 3-minute scrub could be as effective as the 5-minute scrub, depending on the formula of the scrub agent. Immediate and postoperative hand bacterial counts after 5-minute and 10-minute scrubs with 4% chlorhexidine gluconate were compared .The 10-minute scrub reduced the immediate colony count more than the 5-minute scrub. However, the difference between post-scrub and postoperative mean CFU counts was higher for the 10-minute scrub than the 5-minute scrub in longer (>90 minutes) procedures. The study recommended a 5-minute scrub.

In another study scrubs of 2, 4, and 6-minutes duration were compared. A reduction in post-scrub bacterial counts was found in all three groups. Scrubbing for longer than 2 minutes did not confer any advantage. This study recommended a 4-minute scrub for the surgical team's first procedure and a 2-minute scrub for subsequent procedures. Based on the combined data the WHO currently recommends a 5-minutes scrub.

Prior to entry into the prep area.

Don clean scrubs. Scrub tops should be tucked in.

Remove loose jewellery, watches, and rings.

Apply surgical head cover, mask and shoe covers or designated footwear.

Hands should be visibly clean and dry. Fingernails trimmed less than 3mm in length. Fake nails or loose flaking nail polish should be removed.

Remove all hand and arm jewellery and tuck scrub top into pants. Ensure that hair coverings, shoe coverings and surgical face mask are worn. Ensure that a 'social wash' of hands is performed to remove visible dirt from hands and forearms.

Proceed to scrub bay.

Open brush, wet hands and arms, dispense scrub and lather hands and arms.

Clean under nails with nail pick, rinse periodically under running water.

Using nylon side of brush, scrub dorsal surface of all fingernails with bristles (approx 20-30 strokes).

Now switch to sponge side of the brush.

Divide fingers into 4 planes and scrub each sequentially (15 strokes per plane).

Scrub between fingers (webbing) (15 strokes per plane).

Divide hand into 4-6 planes (to ensure complete coverage) and scrub sequentially (15 strokes per plane).

Scrub proximal (towards elbow) and distal (towards the hand) planes of both forearms.

Rinse off from fingers to elbows ensuring that the level of your hands never drop below your elbows.

Collect more scrub from dispenser and lather hands only.

Rinse.

A sterile towel must be used to dry the hands before donning a gown and gloves.

Protocol for surgical scrub with a medicated soap

Wet hands and apply chlorhexidine or iodine soap from the dispenser to hands and forearms up to elbows

Use nail pick to clean under fingernails

Rinse from hands to elbows keeping fingers above the elbows

Apply chlorhexidine or iodine soap from sterile sponge to hands and arms up to the elbows

Start timing. Using the sponge side, scrub each side of each finger, between the fingers, and the back and front of each hand for approximately 2 minutes and each forearm for approximately 1 minute.

Only use the brush side for the fingernails.

The entire scrub should last for 5 minutes.

If a minor break in technique such as bumping the tap occurs. Scrub that area for an additional minute.

Keeping fingers above the elbows, rinse from fingertips to the elbows.

Dry hands and arms with a sterile towel working from the fingertips to the elbows.

Surgical hand preparation with alcohol-based handrubs

Although medicated soaps have been and are still used by many surgical teams worldwide for presurgical hand preparation, it is important to note that the antibacterial efficacy of products containing high concentrations of alcohol by far surpasses that of any medicated soap presently available. In addition, the initial reduction of the resident skin flora is so rapid and effective that bacterial regrowth to baseline on the gloved hand takes more than six hours. Skin irritation and dermatitis are more frequently observed after surgical hand scrub with chlorhexidine than after use of surgical hand antisepsis with an alcohol-based hand rinse.

The hands of the surgical team should be clean and dry upon entering the operating theatre by washing with a non-medicated soap. The activity of the handrub formulation may be impaired if hands are not completely dried before applying the handrub or by the washing phase itself. The application technique has not been standardized throughout the world. The WHO approach for surgical hand preparation requires the six basic steps for the hands as for hygienic hand antisepsis, but requires additional steps for rubbing the forearms. The hands should be wet from the alcohol-based rub during the whole procedure, which requires approximately 15 ml depending on the size of the hands. One study demonstrated that keeping the hands wet with the rub is more important than the volume used. The size of the hands and forearms ultimately determines the volume required to keep the skin area wet during the entire time of the handrub.

The time required for surgical alcohol-based handrubbing depends on the compound used. Most commercially available products recommend a 3-minute exposure, although the application time may be longer for some formulations, but can be shortened to 1.5 minutes for a few of them. The manufacturer of the product must provide recommendations as to how long the product must be applied.

EQUIPMENT CONSIDERATIONS

Our hospital uses a variety of equipment for patients, and it is important to have a system to help determine what level of sterility each item needs to reach before its next use. The system we refer to is Spaulding's classification. It is classifies medical equipment and devices into three classes according to its infection risk during use.

Category of risk	*Application*	*Example of items*	*Process to be used*
Critical	*Entry or penetration into sterile tissue*	*Surgical instruments, surgical implants*	*All items must be purchased sterile or sterilized before use*
Semi-critical	*Contact with intact non-sterile mucosa or non-intact skin.*	*Flexible endoscopes, anesthetic equipment*	*Sterilisation is preferable where possible, otherwise a high level disinfection is required*
Non-critical	*Contact with intact skin*	*Exam tables, blood pressure cuffs*	*Clean as necessary with disinfectant and water*

STERILISATION OF INSTRUMENTS

Complete sterilisation of surgical instruments and any items that might come in contact with the surgical field is a crucial procedure.

Poor sterilisation or inappropriate handling of instruments after sterilisation can result in contamination of sterile tissues during surgery.

Steam sterilisation (i.e. autoclaving) is most commonly used in veterinary clinics. Also available in this facility is plasma sterilisation and Ortho-Phthalaldehyde (OPA) based high level disinfectant

Quality control testing of autoclaves should be performed regularly and documented:

Daily validation:

- Turn autoclave on.
- Run a rapid process cycle with an empty chamber to warm the machine up. Validate the cycle by looking at the cycle printout, and initialling the printout if process ok.
- Run a leak rate cycle with an empty chamber. Validate the cycle by looking at the cycle printout, and initialling the printout if process ok.
- Run a Bowie-Dick cycle with colour change indicator in chamber. Validate the cycle by looking at the cycle printout, and initialling the printout if process ok.
- Then run daily cycles. Validate each cycle by looking at the cycle printout, and initialling the printout if process ok.
- Turn machine off at the end of the day.

Daily validation during use:

- Use external sterility indicators to determine if the item has been processed through the autoclave.
- Use internal sterility indicators to determine if sterility has been reached at the most challenging part of the pack. This is checked after opening the pack but prior to use on the patient.

12 monthly checks:

- The autoclave is serviced every 12 months. At this time, the machine is checked and tested to unsure it reaches the correct temperature, pressure & time parameters.

Sterrad is used on items that have a heat tolerance up to 50 degrees Celsius. Items that are absorbable are not suited. The quality control of Sterrad is as follows:

Daily validation:

- After each cycle is completed, validate each cycle printout by checking that process ok and that the injection pressure has reached the correct parameters of 6 – 14 TORR.

Weekly Validation:

- Biological sterility indicators should be used weekly. These indicators contain bacterial spores, which are the most resistant form of bacteria. After being sterilised, the indicator is submitted for testing to ensure that all of the spores have been killed by the sterilisation process. In human healthcare facilities it is recommended that these indicators are used daily, or at least weekly. Weekly is adequate based on our facility's usage.

Daily validation during use:

- Use external sterility indicators to determine if the item has been processed through the autoclave.
- Use internal sterility indicators to determine if sterility has been reached at the most challenging part of the pack. This is checked after opening the pack but prior to use on the patient.

Flash sterilisation should not be used unless absolutely necessary for emergencies only. Flash sterilisation should never be used for surgical implants. Countertop "cold sterile" disinfectant solutions should not be used for any surgical instruments or implants, as these solutions typically do not achieve true sterilisation of the instruments they contain.

DISINFECTION OF ANAESTHETIC EQUIPMENT

Endotracheal tubes: In human medicine, endotracheal (ET) tubes are typically considered single-use devices, but reuse of ET tubes has become more common with the rising costs of healthcare. These tubes can be effectively re- sterilized between patients, although the physical integrity of the cuffs in particular can be compromised by repeated sterilisation with these methods. These tubes are considered semi- critical equipment, and as such should be subjected to high-level disinfection or sterilisation. In veterinary medicine, it is impractical to discard ET tubes after a single use. Evidence-based guidelines for reuse of ET tubes in veterinary medicine are not available. Nonetheless, at an absolute minimum, ET tubes must be thoroughly cleaned (inside and outside) with hot water and detergent immediately after use to prevent any discharge or debris from drying and forming a biofilm on the device. Tubes should then be soaked in a solution of a quaternary ammonium compound (QAC) or enzymatic cleaning agent, and then rinsed thoroughly and dried prior to being reused. It is important to test the integrity of the cuff

before every use to ensure the device has not been compromised by repeated exposure to the disinfectant.

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

In veterinary facilities

1. All debris i.e. mucous is rinsed off and tube tie is removed
2. The spill valve on the non-rebreathing Bain circuits and Ayers T-pieces are removed before washing as not to damage the filter
3. ET tubes cuffs are inflated
4. Tubes, circuits and rebreathing bags are then soaked in Medizyme (6ml/1 litre water) for 5 minutes
5. Circuit and tubes are the copiously rinsed in water to remove any trace of Medizyme
6. The circuits, ET tubes and rebreathing bags are dried in the warming unit at 58 degrees
    - The Darvall heating circuits are air dried as not to damage the heating coil in the inspiratory limb
7. Any circuits, ET tubes and rebreathing bags that have come in contact with suspected infectious disease are soaked in F10 for 1:500 dilution for 2 minutes before being thoroughly rinsed and dried

The soda lime canister is rinsed and soaked in F10 for 1:500 dilution for 2 minutes before being thoroughly rinsed and dried and refilled with fresh soda lime. If an animal has a known or suspected transmissible respiratory tract infection, filters are available which can be placed between the ET tube and the rest of the anaesthetic circuit in order to help protect the equipment from contamination.

PERI-OPERATIVE ANTIMICROBIALS

Administration of peri-operative (i.e. before, during and after surgery) antimicrobials is an important and complex issue. The goal of peri-operative antimicrobial therapy is to reduce the risk of post-operative infection, while minimizing the negative impact on the patient's natural microflora and the risk of antimicrobial-associated complications such as diarrhoea.

There is currently very little objective information about the need for antimicrobials for specific veterinary procedures, as well as the optimal choice of drug(s), timing and dosages. Antimicrobials are indicated in clean-contaminated, contaminated and dirty procedures. The need for antimicrobial prophylaxis in clean procedures is unclear. In human medicine, antimicrobials are not typically recommended for clean procedures such as arthroscopy, however there are conflicting opinions. Regardless, it is unclear whether recommendations from human medicine should be directly extrapolated to veterinary procedures, because there are

obvious differences in post-operative incision care and patient environment for animals, which may increase the risk of infection. The need for peri-operative antimicrobial therapy for different procedures, particularly clean procedures, requires further research. Concerns with this practice that currently exist include inappropriate timing of administration (i.e. too far in advance of surgery or starting after surgery), excessive duration of therapy, inadequate dosing and inappropriate drug choice.

If peri-operative antimicrobials are used, they should be administered so that therapeutic levels are present at the surgical site at the time of first incision. This typically requires parenteral (i.e. not oral) administration of an antimicrobial approximately one hour before surgery. If the surgical time is longer than two half-lives of the drug(s), then an additional dose(s) should be given during the surgery. In human medicine, it has been shown that starting antimicrobial therapy after surgery is no more effective than not using antimicrobials at all. Typically, antimicrobials are not needed after surgery since the highest-risk time for contamination of the surgical site (i.e. during the surgery itself) is already passed.

SURGICAL SITE MANAGEMENT

PRE-OPERATIVE CARE

Pre-operative management of the surgical site may be very important, but there has been very little research in this area in veterinary medicine.

The goal of pre-operative surgical site management is to eliminate potential pathogens without creating a physical environment that may increase bacterial colonization or infection post-operatively.

In the case of a planned elective surgery, bathing the animal before surgery is reasonable if there is adequate time for the hair coat to dry before the procedure. Owners can be requested to bath their dog the day prior to surgery. They also need to be adequately protected against external parasites. Aseptic technique is improved if the patient is generally clean and parasite free. This is obviously not possible prior to an emergency surgery. However the cleanliness of the patient and the parasite burden needs to be considered. In the cases of a very dirty animal or one with a heavy flea burden, it is advisable to expand the clip site and/or spray the fur surrounding the clipped site with frontline. This all depends if the emergency allows the extra few minutes that this would take.

In humans, it has been suggested that any method of hair removal can be associated with higher SSI rates, but obviously this cannot be avoided for the vast majority of procedures in veterinary medicine. Shaving the surgical site the night before has been associated with higher SSI rates in humans, therefore clipping (not shaving) of the surgical site should only be performed right before surgery. Care must be taken to avoid damaging the skin during this procedure, as abrasions provide sites for invasion and proliferation of opportunistic bacteria. Use of good quality, well-maintained clippers and blades helps to reduce the risk of skin abrasions.

Using a clipper with a #40 surgical blade, generously clip the hair from the area surrounding the proposed surgical site. #50 blades are available and are for use on rabbits and cats with very fine hair. Vacuum loose hair.

If skin lesions around the surgical site are noted before or after surgery, the finding should be recorded and investigated, to determine whether equipment maintenance and/or personnel training need to be improved. Animals should not be clipped in the surgery area/suite itself. A

"prep" area outside of the theatre should ideally be used for this and any other pre-operative procedures.

Animals presenting for elective surgery, found to have evidence of pyoderma or infective skin lesions in the vicinity of the surgical site, should not proceed to surgery. Their skin condition should be appropriately investigated and treated and the animal rescheduled for surgery once the skin irritations have resolved.

Prep one

At any time during the surgical prep, no one is to reach their arms over the surgical prep site.
Exam gloves and a protective nurse's gown should be worn during skin preparation. Prep One should be performed in the surgical prep room.
An initial scrub is done of the surgical site using a 0.5% solution of 4% chlorhexidine (Hexawash) and warm water. The area is to be cleaned with clean swabs. All dirty swabs are to be placed straight into a bin after each scrub.
Initial scrub to remove dirt and hair until swabs appear clean after wipe
When moving patient to surgery suite, the surgery site must not be touched or contaminated. A sterilised gown sleeve may be used to cover limbs that have already been prepped as the patient is moved.

Prep Two:

Perform in surgery suite.
Use sterile pot and swabs. Open using aseptic technique, and pour methylated spirits on the swabs.
Hands should be washed appropriately before donning sterile gloves (use open gloving technique).
Apply methylated spirits in concentric circles starting at proposed incision site, working outwards to clip site margin.
Never go back toward the centre/incision site with the same gauze. Be careful not to contaminate the swab with the hair on the outer of the clip.

Prep Three:

5% Chlorhexidine and 70% methylated spirits solution.
Performed in surgery suite.
Spray directly on the clipped site.
Needs at least 2 minutes skin contact time.
Need to reapply spray at least 2 minutes before draping.

HANGING LEG PREP

Step One
Clip site & vacuum
Bandage lower limb with coflex/rip rap
Apply duct tape over the top of coflex as a waterproof wrap

Step Two
Purse string for approved patients
Place sign "purse string" on patient's head – this is to remind everyone that a purse string has been placed and needs to be removed straight after surgery has finished

Step Three
Complete "Prep One"
Place a clean and sterilised gown sleeve over the limb and attach to leg with a towel clamp
Move patient to surgery suite

Step Four
Remove gown sleeve and clamp distal end of duct tape wrap with towel clamp
Position drip stand next to table, with adjustor handle pointing away from table
Hang clamp from drip stand.
Raise the stand to extend limb to adequate height

Step Five

Complete prep two and three (above). Care should be taken to avoid prep solutions running down from the proximal region of the hanging limb onto the surgical site. This can be avoided by removing excess prep solution from the swabs prior to their use.

Potential problems that need to be avoided include:

- Failure to prepare a large enough area of skin
- Inadequate initial cleaning with soap and water
- Contamination of preparation solutions
- Inadequate contact time with the antiseptic
- Contamination of the area during or after preparation due to improper technique

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

POST-OPERATIVE CARE

Post-operatively, a surgical incision site is highly susceptible to opportunistic infection from the bacteria of the patient's own microflora, from the environment or from hospital personnel. Contact with the surgical incision, particularly with bare hands, should be avoided. Covering or bandaging incisions for a minimum of 24 to 48 hours after surgery has been recommended in humans; this is also a reasonable recommendation in small animals in most situations. Bandage changes should be performed using aseptic technique. Pet owners and handlers should be instructed on how to manage an animal with an incision, and the signs for which to look that may indicate the development of a SSI. There is no objective information about the need to cover surgical incisions for more than 48 hours in veterinary or human medicine, but arguments can be made for both sides. Preventing the animal from licking, scratching or otherwise traumatizing the surgical site is critical. Damaging to the healing incision or the skin around it can result in the deposition of opportunistic pathogens, and make it easier for bacteria to grow in the area.

8. PATIENT CARE AND HANDLING

ISOLATION FACILITIES

The isolation area is dedicated to caring for and housing animals with potentially contagious infectious diseases. An isolation area should allow for complete physical separation of potentially infectious cases, and have areas for performing routine procedures such as bandage changes, thereby reducing the risk of direct or indirect infection of other hospitalized animals or clinic personnel. Ideally, isolation facilities should be in a low traffic location within the clinic.

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

Ventilation should be designed such that movement of air from the isolation room to other areas of the clinic is prevented (i.e. the room should be vented to the outdoors). If this is not readily possible, a HEPA air filtration system should be used for the air leaving the isolation room.

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

When the isolation room is in use by an animal with a potentially contagious infectious disease, prominent signage should indicate that the animal may be infectious and should outline any additional precautions that need to be taken in addition to routine isolation protocols. Access to the isolation room should be limited to the minimum number of essential personnel necessary to provide appropriate patient care.

PERSONAL PROTECTIVE EQUIPMENT AND WASTE IN ISOLATION

All personnel entering an isolation area housing a potentially infectious animal, regardless of whether they plan on having direct contact with the animal, must wear appropriate personal protective clothing. Personal protective clothing consists of a disposable gown, disposable examination gloves, shoe covers, a head cover and P2 masks. These disposable items are disposed of into the clinical waste bin prior to exiting the isolation room.

- Gloves should be discarded after a single use. Hands must be washed immediately after gloves are removed.

- Similarly, gowns should be discarded (if disposable) after a single use. Storing/hanging and reusing a contaminated gown or lab coat inevitably leads to contamination of hands,

clothing and the environment. Therefore, when removed, these items should immediately be placed in the isolation room garbage or laundry bag.

- Eye/nose/mouth protection may be re-used with the same animal if they are not visibly soiled and can be consistently removed without contamination of the inside of the eye wear/mask or the immediate environment. Nose and mouth masks should only be reused by the same person. If the eyewear or mask becomes contaminated with body fluids such as urine or faeces, it should be replaced with a clean article. Designated personal protective equipment must remain in the isolation room.

- Contaminated items and waste alike should be bagged prior to being removed from the isolation area. Articles should then immediately be either discarded or taken to the appropriate area for additional cleaning and disinfection. Waste from an isolation room should be treated as potentially infectious.

- Staff should ensure the removal and disposal of PPE and then replacement of new PPE when moving between patients housed within the isolation ward.

- Staff tending to isolation patients should minimise contact with other young or 'at-risk' patients within the veterinary hospital where possible

PATIENTS IN ISOLATION

Dogs who are housed in isolation should not be walked nor allowed to urinate or defaecate in public areas or areas used by other animals. If a dedicated area for walking is not available and the dog needs to be taken out of the primary isolation area to urinate and defecate, a separate run should be designated for each dog in isolation (i.e. if there is more than one animal in isolation, they cannot all use the same run). The run selected should be as far as possible from runs being used by other animals. The dog should be moved directly to the run by personnel wearing appropriate personal protective clothing. Moving the animal through other areas of the clinic should be avoided as much as possible. Carrying the dog or transporting it on a gurney is ideal in order to minimize the risk of contamination of the floor and clinic environment. The designated run should be prominently labeled and disinfected daily.

If a patient being housed in isolation absolutely must be taken elsewhere in the clinic for essential procedures such as radiographs or surgery, if at all possible this should be done the end of the day, or during a time where there is the least animal and personnel movement in the clinic.

- Appropriate personal protective equipment should be worn by all personnel involved with the procedure.

- Other animals should be kept out of the procedure area.

- The procedure area should be thoroughly cleaned and disinfected as soon as the procedure is completed.

FOOTBATHS AND FOOTMATS

Footbaths or footmats are used to decrease (but do not eliminate) microbiological contamination of footwear. Footbaths are shallow containers containing a disinfectant solution. Footmats are spongy commercial mats covered with a durable, easy-to-clean material that can be saturated with disinfectant. Footmats can increase compliance because they are easier to use, but they are more expensive and more difficult to maintain than footbaths.

Data regarding the need for and efficacy of footbaths and footmats are very limited, and there is essentially no information relating to small animal clinics specifically. It has been shown that footbaths can reduce bacterial contamination of footwear in large animal clinic settings. Although other sources of contamination have been shown to be more significant in infection transmission, footwear and floor surfaces cannot be overlooked in an infection control program in a small animal clinic, because patients so often have extensive direct contact with the floor. Possible problems with footbath or footmat use must also be considered. Footbath or footmat use is almost invariably accompanied by spillage of disinfectant solution; this can create a slipping hazard on smooth floor surfaces, which are typically present in small animal clinics. Certain disinfectants can also damage floor surfaces with prolonged contact.

Footbaths or footmats should be considered when personnel will be walking on a surface that could potentially be more contaminated than the general floor environment, and where spread of this contamination might pose a risk to patients or personnel. The most likely area where footbaths or footmats could be useful would be at the exit of an animal housing area (e.g. dog run) that contains a potentially infectious case, and where clinic personnel will be walking in and out of the potentially contaminated area. The need for routine use of footbaths or footmats in isolation areas where animals are confined in cages is questionable. If footbaths or footmats are used, selection of an appropriate disinfectant is important. The disinfectant should be effective against the specific pathogen(s) of concern, stable in solution, and effective with a relatively short contact time (see Tables 5 and 6). Oxidizing agents such as accelerated/stabilized hydrogen peroxide and peroxygen disinfectants are ideal. The solution should be changed daily, or sooner if gross contamination of the bath/mat occurs.

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

The Veterinary Medical Centre has eliminated the use of footbaths and footmats in lieu of disposable foot covers.

Isolation Procedures at the UQ Veterinary Medical Centre - SAH

The VMC SAH has a dedicated Isolation Ward that is separate from the remainder of the hospital. It has an external entrance that is separate from the hospital entrance. Once inside the Isolation Ward there is a dedicated area for placing belongings and for the placement of appropriate PPE. There is a footbath between this area and the isolation ward which is filled with an appropriate disinfectant.

ALL personnel entering the isolation area must wear the appropriate PPE: disposable gowns, shoe covers, face mask, head cover, and gloves.

Upon exiting the isolation area, all disposable items worn should be discarded in the clinical waste bin provided.

Animals who are housed in isolation are not to be walked, or be allowed to urinate or defecate in areas used by other animals

If an infectious patient requires procedures within the clinic (e.g. x-rays), the patient must be transported on a trolley to avoid contamination of the clinic floor. These procedures should also be performed at a time where there is the least amount of patient and personnel traffic within the clinic (i.e. last procedure of the day). Appropriate PPE must be worn by ALL staff transporting the animal, and the procedure area must be cleaned and disinfected thoroughly after use. Most patients in isolation are managed without moving them into the general clinic for further investigation. The decision to move the patient into the clinic for a procedure is discouraged unless it is deemed crucial in the management of the patient.

Indicators for isolation

The Isolation Ward is used to isolate dogs and cats with infectious diseases that could potentially infect other patients.

These diseases include (but are not limited to):

- Canine Parvovirus
- Feline Herpesvirus and/or Feline Calcivirus (Cat Flu)
- Canine Infectious Tracheobronchitis (Kennel Cough)
- Feline Leukaemia Virus
- Psittacine Beak and Feather Disease
- Psittacosis (Chlamydiosis)

Admission protocols

The receptionist or nurse making an appointment for a coughing dog, a dog with bloody diarrhoea, a sneezing cat, a parrot with feather loss, a parrot with sore eyes and/or sneezing, etc. should request the client to leave the animal in the car on arrival (if safe to do so e.g. stability of the patient, environmental temperature etc).

Explain to the client that their pet may be infectious to other pets and that, for the safety of these other pets, we may need to isolate their pet.

The diagnosis of infectious diseases is made after veterinary examination but if this cannot be made immediately – and where the index of suspicion is high – patients can be admitted to the Isolation Ward prior to a final diagnosis.

Once the decision is made to admit an animal into isolation, the animal must be transported on a stainless steel trolley and not permitted to walk on (or come into contact with) the floor. Any surfaces touched by the animal or coming into contact with body fluids or discharge must be cleaned and disinfected immediately. Staff transporting the animal must be fully gowned and gloved with disposable PPE. The most direct route of entrance should be followed.

Patient contact and movement

- Once an animal is placed into the Isolation Ward, the animal must be housed there for the duration of their stay
- Animals in the Isolation Ward must not be walked outside the Ward
- Samples may be collected from a patient housed in the isolation ward (for e.g. a blood sample) and sent to the laboratory, or run on in-house laboratory equipment. The sample is collected ensuring the least amount of contamination of the sample container. The outside of the container is sprayed with disinfectant and then placed into a clean disposable glove. The nurse/vet that is analyzing the sample should wear gloves at all times when processing the sample. If the sample is being sent to an external laboratory it should be double bagged, and a warning should be placed on the front of the bag to indicate that the sample may be contaminated with an infectious organism.
- Animals who need further work up within the VMC (e.g. radiology, ultrasound) can be brought into the VMC only if the following conditions are met:
    - Wherever possible, procedures within the VMC involving these animals should be the last procedure of the day
    - Personnel handling the animal must be fully gowned and gloved with disposable PPE
    - The animal must be transported on a stainless steel trolley and not allowed to walk on (or come into contact with) the floor
    - Any surfaces touched by the animal or coming into contact with body fluids or discharge must be cleaned and disinfected immediately
- Patients been discharged from the Isolation Ward must not enter the VMC – they are to be taken directly to the client's car on a leash or in a carrier provided by the client

Cleaning, disinfection and waste disposal

Separate cleaning equipment including spray bottles, mops, buckets etc. must be kept for this area within the area and not removed.

All surfaces including bench tops, inside cages, cage doors and food bowls must be sprayed with an appropriate disinfectant. Please refer to the Disinfectant Protocol on the use and handling of disinfectants. If Virkon is used, it must be freshly prepared every 7 days. Alternative commonly used disinfectants in the veterinary clinic are F10 and Oxivir.

Floors must be mopped daily with a disinfectant as appropriate. All organic matter should be removed before the disinfectant is used.

Yellow clinical waste bags must be kept in the room for disposal of bedding and rubbish. Disposable boot covers, face masks, head covers, gowns and gloves that have been worn while in the presence of an infectious patient must be disposed of in a yellow clinical waste bin upon exit.

Patient Bedding – Soiled bedding of infectious patients must be placed in a clinical waste bag for disposal. Bedding is NOT to be moved to the general hospital for washing.

Personnel movement and barrier precautions

No unnecessary personnel are to enter the ward; no more than 4 people are to be in the ward at any one time

Disposable boot covers, head covers, facemasks, gowns and gloves are to be worn at all times whenever there is entry into the isolation ward. These items are not for multiple use, and must be disposed of in a yellow clinical waste bin upon exiting the ante room

Clients may enter the Isolation Ward, but only:

- By appointment
- Under supervision

They must follow the same precautions as detailed above. It is the responsibility of the senior person present to ensure biosecurity is not breached by a client

Supply stocking

The nurse rostered for duty in the Isolation Ward is responsible for stocking supplies in the ward.

Supplies include, but are not limited to, the following:

- Food
- Tinned food
- Hills AD diet
- Liquid food
- can opener
- Consumables
- Syringes and needles
- Thermometer covers
- Fluid bags, administration sets, extension sets, IV catheters
- Bandaging material
- PPE
- Gloves
- Disposable overalls
- Foot covers
- Facemasks
- Head covers

Medications are to be brought into the ward when required, and are not to leave the room.

Stocking levels are to be checked in the morning and then restocking is to occur in the afternoon, unless the need is urgent.

WOUNDS AND BANDAGES

Wound infections can be caused by many bacterial pathogens, some of which can be transmitted between animals or between animals and people. This includes both multidrug resistant (e.g. S. aureus, S. pseudintermedius, enterococci) and susceptible bacteria. Wounds provide a prime site for invasion of opportunistic bacteria such as these. Even wounds that are

not known to be infected should be protected from contamination by veterinary personnel and from the environment to reduce the risk of secondary infection.

- Sterile gloves should be worn for debridement, treatment and bandaging of deep wounds and those involving vital structures. Clean, non-sterile examination gloves are adequate for these procedures if the wound is more superficial.
- Bandages must be kept dry to prevent bacterial strike-though. This means keeping the outside of the bandage as dry as possible, and also including sufficient absorbent material in the bandage itself to prevent discharge from the wound from soaking through the bandage. If the outside of a bandage appears wet, it should be changed.
- Used bandage materials should be considered infectious. Such materials should be placed directly in the garbage and not on the floor, examination table or any other surface. The risk of contamination and spread of any pathogen is likely higher for wounds with a large amount of discharge.
- Wound treatments and bandage changes should be performed in an area that is easily disinfected (e.g. on an examination table). Wound irrigation and lavage should be performed in such a way that the fluid used is contained (e.g. in a sink or tub, or with disposable absorbent material). Bandages should NOT be changed in the kennel/ward area where there is a higher risk of cross-contamination of other patients.
- Hands should be washed thoroughly after changing a bandage. Equipment used for bandage changes (e.g. bandage scissors) should be disinfected between uses.
- Animals with known MRSA or multi-resistant bacterial wound infections are likely to be colonized with these pathogens at other body sites as well (e.g. nose, rectum, intestinal tract), and should therefore be handled with contact precautions and housed in isolation.

FEEDING OF RAW MEAT

Raw meat-based diets for cats and dogs often contain a variety of enteropathogens, including Salmonella spp, Campylobacter spp, Clostridium difficile, Clostridium perfringens, extended spectrum beta-lactamase (ESBL) Enterobacteriacea, and enterohemorrhagic strains of *Escherichia coli* such as O157:H7. It has also been shown that animals fed raw meat diets may shed higher levels of Salmonella and ESBL Enterobacteriacea in their faeces. Raw meat diets and faeces from animals fed these diets may pose a risk to hospitalized animals and clinic personnel, and may contaminate the hospital environment. Therefore, a policy against the feeding of raw meat to hospitalized animals should be in place. Clients who do not wish to have their animal fed a commercial kibble diet could consider cooking the pet's normal diet for the duration of the hospitalization period. However, if it is the opinion of the attending veterinarian that changing an animal's diet from a raw meat diet would adversely affect the animal's health, then the following guidelines should be followed:

- Animals regularly fed raw meat should be housed in isolation and considered infectious. All protocols for handling isolated animals should apply.

- Raw meat should be kept frozen until the day before feeding. It should be thawed in the refrigerator on the bottom shelf in a sealed container.

- Any uneaten meat should be promptly discarded in such a way that it will not attract nor be accessible to insects, vermin or other animals. Significant bacterial growth can occur in any meat that is left out at room temperature, even for a short period of time.

- Any items that come in contact with raw meat (e.g. bowls, storage containers) should be cleaned and disinfected immediately after use.

- Hand hygiene should be strongly emphasized after handling raw meat or any items that have been in contact with raw meat.

ADMISSION OF ANIMALS FROM SHELTERS

Humane societies, animal shelters and similar facilities typically contain transient, stressed populations of animals, large numbers of young animals, sick animals and animals with unknown health and vaccination status. As such, they should be considered high risk from an infectious disease standpoint. Animals admitted from these facilities should be subjected to a high degree of scrutiny. Recommended practices include:

- All animals from such facilities should be examined immediately upon arrival. They should not be allowed to come in contact with other animals in the waiting/reception area.

- If there is an ongoing outbreak of an infectious disease at an animal shelter, admission of animals from the facility for elective procedures should be restricted (i.e. admission for emergencies only). Otherwise, all animals from the facility should be admitted directly to isolation.

- Animals from these facilities should be housed separately from other patients, if possible. Use of a separate ward, separate area of a ward or leaving empty cages between those animals and other patients can be used, depending on the degree of separation required for the diseases of primary concern.

For elective procedures (e.g. spay, neuter):

- All dogs and cats must have received other routine vaccinations (as needed according to geographic region).

    - at least twice if they are more than 14 weeks old, with the most recent vaccine administered at least 2 weeks prior to presentation.
    - All animals must have been dewormed with a broad spectrum anthelmintic at least 7-10 days prior to admission.
    - Animals with abnormalities including, but not limited to, fever, oculonasal discharge,
    - coughing/sneezing, diarrhea and potentially infectious skin conditions should not be admitted for elective procedures.
    - Depending on the geographic region and time of year, flea treatment prior to admission.

9. SAFETY OF CLINIC PERSONNEL

- **Bites and scratches**

In general, veterinarians and animal handlers should be able to recognise behaviour in animals and situations that are associated with an increased tendency for an animal to bite. Professional judgment should be exercised to guide bite prevention practices.
Precautions may include physical restraint or chemical restraint (sedation or anaesthesia) of an animal. Appropriate equipment such as different sizes of muzzles, bite-resistant gloves, halters, rearing bits or a cattle crush should be readily available. Such equipment should also be as easy to clean as possible. Experienced veterinary personnel rather than owners should restrain animals for procedures whenever possible.

Although bites and scratches are considered by many to 'just be a part of practice' every step possible to avoid a bite or scratch should be taken. The following are just some of the steps that should be taken:

- Identify aggressive animals and alert other clinic staff and students of the risk. Place a warning sign on the animal's cage, and flag the animal's patient record of potential/real risk and danger.
- Use physical restraints e.g. muzzles and towels, and/or sedation or anaesthesia as appropriate and in accordance with clinic policy and the patient's clinical condition.
- Plan an escape route when handling any animal, especially one identified as potentially dangerous.
- Do not rely on owners or untrained staff / students for animal restraint.

In the event of a bite or scratch:

- Have someone safely restrain the animal or return it to its cage.
- Wash the site with aqueous chlorhexidine and water immediately.
- Report all bites and other injuries to first aid officer or supervisor for on-line reporting as per UQ guidelines.
- If medical attention is needed contact UQ Security (ext. 53333 or 3365-3333) and advise them of the injury. If there is no response, contact the Gatton Campus Health Service on 5460-1396

Tetanus: All staff should have an initial series of tetanus immunisations, followed by a booster vaccination as recommended by a medical practitioner. In the event of a possible exposure to tetanus, such as a puncture wound, staff and students should be evaluated by their health care provider; a tetanus booster may be indicated.

For details refer to SOP on Y:\SVS\Operations\VTH-Admin\SOP's

- ## Sharps

The basic principles of sharps safety are:

- The person who generates the sharp is responsible for its safe disposal
- Don't pass sharps by hand between people
- Replace sharps containers when ¾ full
- Keep sharps containers out of the reach of children
- Avoid recapping needles unless absolutely necessary.

When it is absolutely necessary to recap needles as part of a medical procedure or protocol, a mechanical device such as forceps can be used to replace the cap on the needle or the one-handed "scoop" technique may be employed (Cornell Center for Animal Resources and Education 2006). This technique involves holding the syringe with the attached needle or the needle hub alone (when unattached) and scooping or sliding the cap, which is lying on a horizontal surface, onto the needle's sharp end.

When injecting live vaccines or aspirating body substances or tissue, the used syringe with the needle attached should be placed in a sharps container.

Needles should never be removed from the syringe by hand. In addition, needle caps should not be removed by mouth.

For detailed SOP, refer to Y:\SVS\Operations\VTH-Admin\SOP's

- ## Sharps Safety for Clients

- Periodically, provide an approved sharps container or give clients clear instructions regarding how to obtain one.
- Ensure owners may be required to treat their animals at home with injectable medications (i.e. insulin, subcutaneous fluids). In these situations, it is the responsibility of the attending veterinarian to:
    - Provide (and document) training on how to handle sharps, including injection and disposal practices.
    - that the client is able to safely handle and dispose of sharps.
    - advise clients that the sharps container should be returned to the clinic for disposal when 3/4 full, and exchanged for a new container (if necessary).
- Used sharps are considered biomedical waste in veterinary practices. Dispose of used sharps containers in accordance with regulations.

For SOP refer to Y:\SVS\SVS-OHS-Public\VTH\Handling Medical Waste and Sharps.

- **Aggressive and potentially aggressive patients and the role of Reception Staff**

Purpose

To ensure all Receptionists are aware of the reasoning and procedures in handling a potentially aggressive patient in the waiting room to protect UQ staff and students, our clients and patients.

Procedure

Incidences may arise when a client may be exposed to other clients' aggressive animals in the VMC, for example by a child who may go and pat a dog in the waiting room.

If a patient e.g. a dog has bitten a child while in the waiting room, it is highly possible that ultimately it will be VMC and UQ which will be legally liable. Even though the owner of the dog was in charge of the dog as well as the child being accompanied by an adult once they are on our premises (carpark and building), we are technically and legally responsible for the incidents and accidents that took place on our premises. Our responsibility increases and the outcome worsens if a dog was known to VMC staff to bite i.e. it has a 'known propensity to commit a dangerous act'.

It is therefore important that, whenever a dog exhibits a potential to bite or any aggressive behaviour, we document it clearly in RxWorks as a client note which pops up upon opening of the client's screen.

Upon arrival, once this was identified, reception staff should escort the client directly to an available consultation room and ask to wait in there. Staff working with the animal should discretely make students aware for appropriate behaviour and animal handling during procedures. While human safety is of paramount by taking all reasonable steps to eliminate incidents and injuries, it is also very important that due to other common Laws and Acts, owners should not be made uncomfortable.

For SOP refer to Y:\SVS\SVS-OHS-Public\VTH\Aggressive and potentially aggressive patients and the role of Reception Staff.

- **Guidelines and the procedures to follow: In case of post contaminated sharps incidents/injuries involving human blood (whether indirect or direct)**

Purpose
To provide staff with guidelines when human to human blood transmission occurs.

Procedure
Working with human or animal blood products, tissue and body fluids increases the risk of a worker acquiring a blood borne infectious disease. Some blood borne infections do not have a vaccination available to prevent transmission of disease, therefore all blood and body fluids should be handled as if they are potentially infectious.

First –aid procedures (also seek help from first-aid officer on-site):

- Bleeding should be encouraged but squeezing and rubbing the affected site should be avoided.
- Puncture wounds or cuts should be washed with soap and water.
- Splashes into the eye should be flushed using an eye wash fountain or saline with eye open for at least 30 seconds.
- If splashing to the face occurs then gentle washing and rinsing several times with water should be used for the skin, nose and mouth.

Following above actions:

- Seek advice from a medical practitioner immediately: UQ Health Services (Gatton Campus) 5460 1396, hospital (e.g. Ipswich or Gatton hospitals) or from your own GP. For incidents involving human to human blood or body fluid exposure, a risk analysis of blood borne virus transmission must be undertaken on the information available of the type and the degree of exposure and the amount and type of infectious material involved. Refer to Tables 1 and 2.
- The medical practitioner will organise referral to an infectious disease specialist if the injury is assessed as high risk for any blood borne virus infection as prophylactic treatment may be required as soon as possible after such an injury. Refer to Qld Health Guideline, Management of occupational exposure to blood and body fluids 2017 https://www.health.qld.gov.au/__data/assets/pdf_file/0016/151162/qh-gdl-321-8.pdf The incident must be reported to (1) the supervisor or the VMC representative; (2) to UQ Occupational Health and Safety Division occupational nurses **3365 4883** ohna@uq.edu.au, and (3) also reported on-line in the http://www.uq.edu.au/ohs/index.html?page=141331 and choose "UQ Safe – Incident".

  For SOP refer to Y:\SVS\SVS-OHS-Public\VTH\Human to human blood transmission.doc

SOP for Reception Staff: Making an appointment for venomous reptiles, SAH, VMC

Making the appointment

If a client calls for an appointment with a venomous reptile, the following actions are to be taken by the receptionist:

1. Ascertain the species of the snake, including the scientific name. This is needed to avoid confusion over the species involved.
2. Ascertain that the handler has a restricted wildlife license, allowing them to keep venomous reptiles. Ascertain the handler's experience in handling venomous reptiles
3. Ensure the handler has the equipment needed to safely transport the snake, including heavy duty canvas bags and tubes to restrain the snake, handling tongs and other equipment.
4. Allow a minimum of 60 minutes for the appointment. Venomous reptiles cannot be hurried.
5. Notify Dr. Bob Doneley (ext. 50980; 0418 987202) or Gary Fitzgerald (ext. 50400; 0423933074) before confirming the appointment. Appointments are not to be made with other clinicians.
6. Appointments for venomous snakes and reptiles must only be made during business hours regardless of a trained staff (see below for who they are) on duty or not after-hours.

Wildlife cases

In the event of a wildlife carer or a member of the public bringing in a venomous (or potentially venomous) snake either in-hours or after-hours for emergency treatment, the following procedures are to be followed:

a. Do not attempt to remove the snake from its container or attempt to examine it.
b. Place it in a plastic container with a lockable lid (these can be found in the exotics ward (1023), or the carer may have brought it with them)
c. Tape the lid closed to prevent accidental opening of the container
d. Place the container in a cage, with a large sign "CAUTION, VENOMOUS SNAKE. DO NOT OPEN" on the front of the cage
e. Contact Dr Doneley (ext. 50980; 0418 987202) or Gary Fitzgerald_(ext. 50400; 0423933074) for further instructions

In case of snake bite, call UQ SECURITY on 3365-3333 and advise you are calling from GATTON CAMPUS, Veterinary Medical Centre, Building 8156 and the nature of the emergency – in this case, venomous snake bite.

Also refer to: Y:\SVS\Operations\VTH-Admin\SOP's

Diagnostic specimen handling

- Urine from animals with suspected urinary tract disease, and all faeces, aspirates, and swabs

should be treated as potentially infectious material. Protective outerwear (e.g. lab coat) and disposable gloves should be worn when handling these specimens. Gloves should be discarded and hands washed immediately after handling these items. Care should be taken to avoiding touching clean items (e.g., microscopes, telephones, food) while handling specimens or before glove removal. A separate refrigerator should be used for diagnostic specimens, which should be cleaned on a regular basis.

- A designated area of the clinic should be used for specimen processing. This should be separate from treatment and surgery areas so as to decrease the risk of contamination of these areas. After processing a specimen, materials should be disposed of or stored properly and promptly.

- Specimen processing areas should be cleaned and disinfected immediately after use.

- Samples from animals with suspected or known infectious diseases should be disposed of as infectious waste.

- Leak-proof plastic containers should be used for specimen storage in a designated refrigerator which does not contain food, vaccines or medications of any kind. Contamination of the outside of sample containers should be avoided. If the outside of a container becomes contaminated, it should be cleaned and disinfected prior to storage.

- Sharps such as microscope slides and glass pipettes should be disposed of in approved sharps containers.

For SOP on samples transport refer to Y:\SVS\SVS-OHS-Public\Sample transport

• Dental Procedures

PPE for dental and obstetric procedures
http://www.ava.com.au/sites/default/files/AVA_website/pdfs/Resource%207%20-%20PPE%20for%20dental%20and%20obstetric%20procedures.pdf

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

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

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

Vaccination of personnel

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

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

Q Fever: Veterinary students are expected to be immunologically protected against Q Fever. For further information see: http://www.uq.edu.au/vetschool/requirements.

Influenza: Human influenza is a common and highly transmissible disease, even though it is not transmissible to companion animals. Infected veterinary personnel can rapidly infect their colleagues and veterinary clinics could act as sources of community infection if infected employees are present. It is reasonable for veterinary clinics to recommend annual influenza vaccination of all personnel to ensure that personnel have time to visit their physician or a vaccination clinic for this purpose. Employees should also be encouraged to stay home if they are ill.

Training and education of personnel

Personnel training and education are essential components of an effective infection control program. All personnel, including temporary lay personnel, kennel staff, students and volunteers, should receive education and training about injury prevention and infection control during their initial orientation and periodically thereafter. Additional training should be provided as recommendations change or if problems with infection control practices are identified. Training should emphasize awareness of the hazards associated with individual work duties, and prevention of zoonotic disease exposure. Staff participation in training should be documented by the infection control practitioner (ICP). A list of additional electronic and print resources that may be useful for training purposes can be found under References.

All personnel should receive education and training about injury prevention and infection control.

Client education

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

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

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

- What disease is suspected or has been diagnosed
- How the disease is confirmed, if necessary
- How the disease is transmitted
- Risks to members of the household
- Risks to other in-contact individuals (e.g. elderly grandparents who live elsewhere)
- Risks to in-contact pets
- Symptoms in humans
- Clinical signs in animals
- How to prevent disease transmission from the pet to people and to other pets
- How the disease is treated in animals
- Public health enforcement issues such as quarantine, submission of tissues to labs, etc.
- Circumstances under which the client should seek medical attention, if applicable

Client visitation

Given the strong bond between owners and their pets, it is understandable when clients wish to visit their hospitalized pets. However, animals carrying transmissible infectious pathogens pose a potential risk to other animals at the clinic and at the owner's home, as well as to the clinic employees, the owner and other household members. As a policy, clients should not be allowed to visit animals that are considered potentially infectious. Under extenuating circumstances, such as an animal whose condition is imminently life-threatening, owners may be allowed to visit their animal, but the use of proper personal protective equipment should be demonstrated to the clients and all infection control procedures should be followed, as for clinic personnel involved in the animal's care.

Client education is the responsibility of the entire practice team.

As a policy, clients should not be allowed to visit hospitalized animals carrying any suspected infectious disease.

Clinic pets

It is currently common for veterinary clinics to have resident animals. From an infection control perspective, these animals pose a potential risk for disease transmission, and from the health perspective of the clinic pet itself. Clinic animals that have free access within the clinic could be sources of pathogen transmission. Uncontrolled access to waiting room areas could result in a large number of contacts, with the corresponding potential for pathogen transmission. Although there are no objective data quantifying the risks to patients, people or clinic animals themselves, the theoretical risks and lack of a real need for clinic pets indicates a need for consideration of the cost- benefit of keeping clinic pets. Based on the potential risks, it is recommended that veterinary clinics do not keep such animals, and every attempt should be made to adopt out any existing pets.

While suboptimal from an infection control standpoint, if a clinic has a clinic pet, the following recommendations should be considered. The clinic pet should not have access to any patient

treatment areas, patient housing areas, examination rooms, isolation, surgery or the patient waiting area. It should not be allowed to wander freely through the kennel/ward areas where it could cross-contaminate kennels. The animal should have a dedicated food and water bowl, litter box, toys, etc. The pet must also receive regular health checks and have an appropriate vaccination, deworming and external parasite control program. Clinic pets, particularly cats, should not be allowed to have unsupervised outdoor access because of the higher risk of exposure to (and subsequent shedding of) pathogens such as Salmonella and Toxoplasma from hunting birds and rodents.

Vector control

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

- Examination of animals upon arrival for ectoparasites such as fleas, and treatment with an adulticidal antiparasitic medication prior to admission if ectoparasites are detected.
- Storing food and garbage in metal or thick plastic containers with tight-fitting lids.
- Prompt disposal of food waste and other material (e.g. feces) that may attract rodents or insects.
- Sealing potential pest points-of-entry into buildings. Common methods include the use of caulk, steel wool or mesh wire under doors and around pipes.
- Installation and maintenance of window screens to prevent entry of insects into buildings.
- Elimination of potential rodent nesting sites (e.g. clutter).
- Removal of standing water (e.g. empty cans, clogged gutters) outside buildings that can otherwise serve as breeding grounds for mosquitoes.

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

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

Reportable diseases

Certain diseases are immediately reportable to regulatory bodies, often at the time the disease is suspected but still not diagnosed. These diseases vary between countries and tend to focus on exotic pathogens and those of significant zoonotic concern (e.g. rabies). Every veterinary clinic should have a list of reportable diseases prominently displayed in an area easily accessible to clinic personnel. The clinic's Infection Control Manual should clearly state the required reporting procedures, including contact numbers for the appropriate animal health and/or public health authorities.

https://www.daf.qld.gov.au/animal-industries/animal-health-and-diseases/notifiable will direct you to the list of Qld Notifiable Diseases.

If you suspect an animal disease listed below or a notifiable disease incident, you must report it to Biosecurity Queensland on 13 25 23 or the Emergency Disease Watch Hotline 1800 675 888.

- ## Queensland's notifiable diseases as of 20 April 2017

- African horse sickness
- African swine fever
- anaplasmosis, if the disease occurs outside a cattle tick infected zone
- anthrax
- Aujeszky's disease
- Australian bat lyssavirus
- avian influenza
- avian mycoplasmosis (*M. synoviae*)
- avian paramyxovirus
- babesiosis, if the disease occurs outside a cattle tick infected zone
- bluetongue (clinical disease)
- Borna disease
- bovine virus diarrhoea type 2
- brucellosis (*B. abortus, B. suis, B. canis and B. melitensis*)
- camelpox
- cattle tick (*Boophilus microplus*) infestation, if the disease occurs outside a cattle tick infected zone
- Chagas disease (*T. cruzi*)
- classical swine fever
- contagious agalactia
- contagious bovine pleuropneumonia (*Mycoplasma mycoides mycoides* small colony type)
- contagious caprine pleuropneumonia (*Mycoplasma capricolum*)
- contagious equine metritis (*Taylorella equigenitalis*)
- Crimean-Congo haemorrhagic fever
- Cysticercus bovis (*Taenia saginata*)
- devil facial tumour disease
- dourine (*Trypanosoma equiperdum*)
- duck virus enteritis (duck plague)
- duck virus hepatitis
- East Coast fever (*Theileria parva*)
- encephalitides (tick borne)

- enzootic abortion of ewes (*Chlamydophila abortus* and *Chlamydia psittaci* serotype 1)
- enzootic bovine leucosis
- epizootic haemorrhagic disease (clinical disease)
- epizootic lymphangitis (*Histoplasma capsulatum* var.*farciminosum*)
- equine encephalomyelitis viruses (eastern, western and Venezuelan)
- equine encephalosis
- equine herpes virus 1 (abortigenic and neurological strains)
- equine infectious anaemia
- equine influenza
- equine piroplasmosis (*Babesia equi*, *Babesia caballi* and *Theileria equi*)
- equine viral arteritis
- foot and mouth disease
- footrot in sheep (*Dichelobacter nodosus*)
- fowl typhoid (*Salmonella gallinarum*)
- Getah virus infection
- Glanders (*Burkholderia mallei*)
- Goat pox
- H1N1 swine influenza
- haemorrhagic septicaemia
- heartwater (*Ehrlichia ruminantium*)
- Hendra virus infection
- infectious bursal disease (hypervirulent and exotic antigenic variant forms)
- infectious laryngotracheitis
- Japanese encephalitis
- Jembrana disease
- Johne's disease (Mycobacterium avium paratuberculosis)
- leishmaniosis of any species
- louping ill
- lumpy skin disease
- lyssavirus other than Australian bat lyssavirus
- maedi visna
- Malignant catarrhal fever (wildebeest associated)
- Mediterranean theileriosis (*Theileria annulata*)
- Menangle virus
- Nairobi sheep disease
- Newcastle disease (virulent and avirulent)
- Nipah virus
- porcine cysticercosis (*C. cellulosae*)
- porcine enterovirus encephalomyelitis (Teschen)
- porcine myocarditis (Bungowannah virus infection)
- porcine reproductive and respiratory syndrome
- post-weaning multisystemic wasting syndrome
- Potomac fever
- pullorum disease (*Salmonella pullorum*)
- pulmonary adenomatosis (Jaagsiekte)
- rabies
- Rift Valley fever
- rinderpest
- salmonella
- screw-worm fly - New World (*Cochliomyia hominivorax*)
- screw-worm fly - Old World (*Chrysomya bezziana*)
- sheep pox
- sheep scab (*Psoroptes ovis*)

- surra (*Trypanosoma evansi*)
- swine influenza
- swine vesicular disease
- transmissible gastroenteritis
- transmissible spongiform encephalopathies (bovine spongiform encephalopathy, chronic wasting disease of deer, feline spongiform encephalopathy, scrapie)
- trichinellosis (*Trichinella spiralis*)
- turkey rhinotracheitis (avian metapneumovirus)
- trypanosomiasis
- tuberculosis (mammalian or avian)
- tularaemia
- vesicular exanthema
- vesicular stomatitis
- warble-fly myiasis (*Hypoderma* spp.)
- Wesselsbron disease
- West Nile virus infection – clinical

Zoonotic disease transmission

On page 35 of http://www.ava.com.au/sites/default/files/Guidelines-for-veterinary-personal-biosecurity-2017-FINAL.pdf will take you to a comprehensive list of Zoonotic diseases of importance to Australian veterinarians.

Sources of zoonotic diseases include animals or environments contaminated by animals. Pathogens may be transmitted to humans directly from the animal via blood or other body substances or indirectly from the animal's environment.

- Routes of transmission:
  - Contact transmission
  - Droplet transmission
  - Airborne transmission
  - Vector-borne transmission

Pregnancy

During pregnancy, women experience physiologic suppression of cell-mediated immunity, increasing their susceptibility to certain infections. These include toxoplasmosis, lymphocytic choriomeningitis virus infection, brucellosis, listeriosis, Q fever, leptospirosis and Chlamydophila psittaci. Vertical transmission of certain zoonoses may result in abortion, stillbirth, prematurity or congenital anomalies. Measures to reduce risk from infection with these pathogens will vary depending on individual circumstances, but may include:

- avoiding jobs such as obstetrics due to the contact with birth fluid
- avoiding contact with young cats, cat faeces or raw meat to lessen the chance of contracting Toxoplasma

In Australia pregnant women are not routinely screened to check their antibody titre against Toxoplasma due to the complexity of interpreting positive results. Employers should ensure that there are safe systems of work to protect the health and safety of pregnant workers, and provide pregnant workers with information about relevant zoonoses and associated risk controls.

Employees who are pregnant or who have immune dysfunction should discuss their status with the practice manager or owner so the practice can provide appropriate workplace accommodations to protect them. The use of infection control measures and personal protective equipment will reduce the risk of infection. In some cases, it may be advisable to consult the employee's healthcare provider (with the person's consent) or an infection control, public health or occupational health specialist in managing the zoonotic disease risk (Grant and Olsen 1999). Employers must abide by state and federal laws that protect pregnant women and persons with disabilities. The employee should be assured that confidential information will not be disclosed to others.

Immunocompromised personnel

Immune deficiencies may put veterinarians and staff at increased risk for acquiring zoonotic infections (Centers for Disease Control and Prevention 2009). Additionally, immunocompromised personnel are more likely to develop serious complications from infections. Immune deficiencies may result from underlying medical conditions (e.g. HIV/AIDS, diabetes mellitus, asplenia, pregnancy, certain malignancies), therapy for a variety of conditions (e.g. steroids, chemotherapeutic and immunosuppressive agents, radiation) or may be congenital.

Immunocompromised employees and their supervisors should be aware of the following workplace encounters that may result in exposure to zoonotic pathogens:

- Processing laboratory samples.
- Direct patient care, especially with the following high risk animals:
    - Young animals (ruminants prior to weaning, dogs and cats less than six months of age)
    - Animals with diarrhoea
    - Parturient animals
    - Stray or feral animals (especially predators of rodents and wildlife
    - Animals fed raw meat diets
    - Reptiles or exotic, imported species
    - Animals housed in crowded conditions (such as shelters)
    - Unvaccinated animals or those with untreated internal or external parasites.

10. UQ Veterinary teaching hospital and small animal clinic: Infectious Diseases in dogs and cats

Procedure:

Infectious Diseases in dogs and cats and the role of Reception

Purpose

- To ensure all Receptionists are aware of the more common infectious diseases that may be presented to UQ VETS SAH and the appropriate advice and information that needs to be extracted and / or provided to clients attending with their animal/s.
- By reception being aware of the basic clinical signs of infectious diseases, they can play a pivotal role in advice given clients of steps to minimise the spread of these diseases within the VMC.
- Primary diseases of interest are Parvo virus, Kennel Cough and Cat Flu

Procedure

Parvo

- Parvo virus is prevalent in the Gatton area and clinical signs of this virus may include vomiting, diarrhoea (with or without blood and even looking like raspberry jam) and generally unwell and depressed in appearance.
- The virus is spread by faeces from infected dogs and is a particularly tough virus and can live for very long periods in the ground.

Vaccinations and puppies.

- The UQ VMC requests that all new puppy owners have their puppies at home for 7 days before they are presented to the VMC in for their first vaccination.
- Owners can be informed that this above request is to ensure their puppy is healthy at time of vaccination due to the increased risk of adverse effects of vaccination if they are unwell.
- Note:- the VMC's intention is to help ensure that if they have contracted parvovirus they will be showing clinical signs by this point and minimise any further potential contamination within the VMC.
- Delays in vaccination may have an impact upon social growth of the puppy during their critical stages of behavioural and social development and there is no intention of impacting this development.

Questioning clients regarding vaccination status

- Parvo cases will unavoidably be presented no matter what checks and balances are put in place
- Reception staff are to confirm with all new clients if their dog/s have been vaccinated. Note that vaccination may not give 100% protection and vaccination status can be misleading.
- **If unvaccinated** any dog showing signs of vomiting or diarrhoea- the animal should be left outside in the clients' vehicle until a vet has assessed them.

- **If vaccinated**- we ideally need to further clarify this information (ie did they have all their puppy vaccinations, have the received booster vaccinations as adults) and request that they bring the vaccination certificate with them allowing staff to review prior to dogs entering the VMC.
- If dogs of any age have only had their puppy vaccinations and they are showing signs of vomiting/diarrhoea- these animals should also be assessed by a vet outside first.

Canine Cough or Kennel Cough

- Kennel cough has been likened to sound like a dog trying to cough up something stuck in their throat. They may also be sneezing and/or have nasal discharge.
- Kennel Cough has always been a hard infectious disease to keep out of any Veterinary clinic as owners become very worried when their pets are coughing and may even think their animal is choking.
- Being as aerosol disease, containment can represent a challenge to clients and Veterinary Clinics alike
- Any dog of any age who is coughing (even if the owner says they have something 'stuck in their throat') should be requested to be left outside for assessment UNLESS they have been seen by another vet an referred over for a foreign body.

Cat Flu

- Any cats being presented for sneezing +/- watery eyes may be suffering from Cat Flu which is a highly contagious infections disease amongst cats which is spread via sneezing or commonly the moisture film on hands
- Clients should be advised to cover the cat's cage when bringing into the clinic to reduce transmission of the virus/s.
- Cats should be moved directly to a Consult room when they arrive and preferably access the VMC through the side door in the waiting room - down near the Internal Medicine room.

Spread of Disease

- Communal areas and communal food/drink containers assist with the spread of disease.

Also refer to Y:\SVS\Operations\VTH-Admin\SOP's

Virulent Systemic Feline Calicivirus (VS-FCV)

Background Information

- Feline Calicivirus is a common infection in cats caused by multiple different strains of calicivirus and is capable of causing flu-like symptoms and more severe disease in some cats.
- A particularly virulent strain of FCV, called Virulent Systemic Feline Calicivirus, recently emerged. This strain is capable of causing severe generalised disease through severe vasculitis and ulcerations by epithelial cell cytolysis (Pesavento et al, 2004).
- UQ VETS had a confirmed case of VS-FCV in October 2017

VS-FCV Disease

- **Virology**
  - All FCV strains are potentially pathogenic and need to be managed collectively
  - VS-FCV is a recognised distinct strain, but has no diagnostic clinical or virological features that differentiate it from other strains. Vasculitis and associated pathology is suggestive of the VS strain
  - RT-PCR best used for Dx of FCV, but won't differentiate the VS strain. No reliance on PCR for Dx – need to combine with clinical and epidemiologic signalment
  - Un-enveloped virus: more resistant to environmental exposure and disinfection methods.
  - High mutation rate, leading to the development of virulent strains and resistance against vaccines.
  - Can be associated with co-infection – complicates the clinical presentation and management, and can confuse regular strains for VS. Other infections (particularly panleukopaenia, FHV-1, *Mycoplasma felis*, *Chlamydophila felis*, and *Bordetella bronchiseptica)* need to be ruled out
  - Incubation period: 2-10 days
  - Persistence in the environment at room temperature for up to 4 weeks.
    Longer persistence in cold conditions
  - Caliciviruses are typically very species specific and do not represent a risk to people or other species of animals

- **Risk profile:**
  - Shelter cats are particularly high risk
  - Other intensively managed facilities, e.g. catteries, breeders, clinics
  - Kittens appear to be more frequent shedders
  - Clinical recovery and/or lack of symptoms does not mean the cat is not potentially shedding virus, although asymptomatic cats will generally excrete less virus
  - F3 vaccine not completely protective for disease and not protective for carriage
  - Stress an underlying risk factor: co-infection, crowding, social, nutritional, co-mingling, poor hygiene, poor ventilation, etc.
  - Carriage of FCV strains (+/- FS variant) common in all cats
  - Shedding can persist up to 4 months in cats recovering from clinical infection

- **Transmission:**

- Highly contagious: cats hospitalised for more than 12 hr with an infected cat or from the same household had more than 90% chance to be infected (Hurley et al, 2004)
- Droplet. Mainly URT, but potentially any excretion
- No true aerosolisation, but possible airborne spread <1m
- Cat-cat contact
- Fomites: especially hands, but including clothing, equipment
- Premises surfaces: floors, benches, cages
- Possibly also on cat's fur – grooming, excretion

- **Clinical Signs**
  - Fever
  - Anorexia
  - Limping
  - Oral ulcers
  - Upper respiratory signs
  - Lower respiratory signs going from tachypnoea to dyspnoea
  - Oedema of face and limbs
  - Ulcerative pododermatitis
  - Icterus
  - Bleeding tendencies with melena, petechiations
  - Sudden death
  - Mortality rate: Case fatality of 40%, ranging from 25 to 70% depending on strain and population described. The mortality rate tends to be higher in older cats (>1 yr old) than kittens (< 6 month old) with respective rates of about 60% and 15% (Hurley et al, 2004).

- **How to diagnose VS -FCV?**
  - PCR from oropharyngeal swabs or other fluids like pleural effusion
  - Virus isolation from oropharyngeal swabs or tissues sampled
  - Genetic sequencing can be done to identify specific strains
  - Serum virus neutralizing titres
  - IHC in tissue samples

- **What can work up of these cases identify?**
  - Pleural effusion and most likely a modified transudate with pyogranulomatous component which can make the disease appear similar to FIP initially
  - Increased liver enzymes and bilirubin due to viral liver damage
  - Increased CK
  - Increased glycaemia due to associated pancreatitis and pancreatic necrosis
  - Decreased albumin
  - Neutrophilia common on CBC
- **Treatment**
  - There is unfortunately no specific treatment, it is mainly supportive care
  - Broad spectrum antibiotics (doxycycline or amoxyclav suggested) for secondary bacterial infection
  - Nutritional management through feeding tube and enteral nutrition
  - Intravenous fluids to be used with caution due to the vasculitis component of the disease and risk of aggravating respiratory symptoms of pulmonary oedema; favour the enteral route for rehydration if dyspnoea or tachypnoea present
  - Interferon has been reported to be helpful in some case but the literature remains controversial on their use. They are not usually associated with marked side effects. It has been suggested to use them in the more severe cases

- Sometimes glucocorticoids are needed to help with a potential immune-mediated component associated with the viral infection; to be used with caution and on a case by case basis not considered gold standard for treatment
- Gastrointestinal supportive care with anti-emetics, anti-acids, sucralfate for the GI ulcers
- Plan to be adjusted with each specific cases as they might present with various combinations of the symptoms listed above

- **Is vaccination protective?**
  - No the current vaccine does not really protect against the virulent strain unfortunately
  - Interestingly, young unvaccinated animals tends to suffer from the mild form whether adult and vaccinated cats could be more likely to suffer from the more severe form

Control Protocol Elements

- **Suggestions on how to manage incoming cases if indicated:**
  - Ensure all staff are aware of VS-FCV
  - All cats considered potentially at risk for carriage or transmission
  - Suspect cases or cats in contact with known carriers triaged to a separate isolation facility
  - All other cats dealt with case-by-case as individuals:
    - Strict decontamination of consult room and equipment between cats
    - Hand hygiene and personnel decontamination between cats
    - Admitted cats: quarantine period of ~7 days with isolation/barrier methods from other admitted cats as much as possible. Careful monitoring for early clinical signs
    - Minimise admissions: advocate home or similar care where possible

- **Isolation of infected and suspect cats:**
  - Separate premises: physical distance, doors, etc. provide barriers to transmission
  - Dedicated equipment and clothing (e.g. scrubs, disposable aprons, boot covers)
  - Separate management of bedding, laundry, feed, waste
  - No clearly defined long term isolation period for recovered cats:
    - Up to 3 months likely for severe disease in vaccinated cats
    - Recovered cats: longer term housing? Fostering?
    - Vaccination will not reduce carriage or shedding

- **Movement control and workflow management:**
  - Of all cats, not just infected/suspect
  - Cats only to be managed by specified staff who do not contact other cats or are trained to manage VS-FCV
  - Physically contact healthy cats before infected/suspect cats
  - Identify VS-FCV ‘clean’ and ‘dirty’ areas
  - Manage staff and animal work flows to limit potential passage of infected individuals, or contaminated materials or humans, through or into ‘clean’ areas
  - No hospital access for non-essential activities or personnel

- **Personnel:**
  - Hand hygiene: based on “5 moments” principles and effective technique

- Use of disposable gloves, gowns, shoe covers for higher exposure risk activities, e.g. with infected cats
- Use of hand sanitisers: wash and alcohol gel stations readily accessible and functional
- Strict use of dedicated clothing (e.g. scrubs) whilst at the hospital
- Staff personal decontamination on leaving the hospital:
  - Hand hygiene
  - Removal of work dedicated clothing: scrubs, etc.
  - Decontamination of equipment (e.g. personal stethoscopes, notepads, footwear, phones, computer keyboards)
- Advised not to be in contact with cats for 48 hrs after hospital contact where possible

- **Decontamination:**
  - Initial cleaning of gross contamination
  - 'Dirty' areas: Rigorous disinfection of all surfaces and equipment in contact with infected/suspect cats
  - 'Clean' areas: strict adherence to routine decontamination protocols, with a recommended increase in frequency of application until the case cluster resolves
  - Recommended disinfectants:
    - Peroxygens: potassium peroxymonosulfate (e.g. Virkon), accelerated peroxides
    - Halogens: hypochlorite (bleach), iodine
    - Accelerated hydrogen peroxides e.g. Oxivir
    - Alcohols: 70% v/v ethanol or propanol
  - Ideally, have 10 min contact time for any disinfectant used. Where not practical, maximise contact.
  - General chemical disinfectant protocols: correct concentration, stock vs working solutions, use of fresh solutions with clear re-constitution guidelines, minimise organic matter contamination

Additional Information

How many other outbreaks described in the literature?

- First recognised outbreak in 1998 in Northern California where it took the name of haemorrhagic fever
- Similar outbreaks in reported across several states across the USA
- Outbreak described in England in 2003 and then several more in Europe.
- Outbreaks first reported in Sydney in Dec 2016/Jan 2016. These might not have involved the same strain as the cats involved appeared not to suffer of systemic signs as markedly as the ones described in our local area. (Hughes, 2016)

Did outbreaks share similar features?

- Most often the case, the index case was a hospitalised shelter cat
- Adult vaccinated cats were affected predominantly and kittens appear to have less signs
- Spread is rapid through fomites and affected client cats and vet staff cats
- Often the spread was limited to the one clinic or shelter and there was no spread associated in the community
- Outbreak resolves on their own within 1 to 2 months usually, although nobody know how and why

- No reported transmission of disease from fully recovered cats, despite them still shedding the virus

Useful references

- Coyne et al. Lethal outbreak of disease associated with feline calicivirus infection in cats. Vet Record 2006; 158:544-550.
- Hughes D. Virulent feline calicivirus (FCV) in Sydney's inner west. Centre for Veterinary Education 2016; 284:25-29.
- Hurley et al. An outbreak of virulent systemic feline calicivirus disease. J Am Vet Med Assoc 2004; 224(2):241-249.
- Hurley & Sykes. Update on feline calicivirus: new trends. Vet Clin Small Anim 2003; 33:759-772.
- Pedersen et al. An isolated epizootic of hemorrhagic-like fever in cats caused by a novel and highly virulent strain of feline calicivirus. Vet Microb 2000; 73:281-300.
- Pesavento et al. Pathologic, immunohistochemical and electron microscopic findings in naturally occurring virulent systemic feline calicivirus infections in cats. Vet Pathol 2004; 41:257-263.
- Radford AD et al. Feline calicivirus infection. ABCD guidelines on prevention and management. J Fel Med Surg 2009; 11:556-564.

Related policies

Refer to Y:\SVS\Operations\VTH-Admin\SOP's\Internal Processes\SAH Specific\Virulent Strain - Feline Calcivirus.docx

- **Brucellosis in dogs guidelines for veterinarians**

Ref: http://www.dpi.nsw.gov.au/biosecurity/animal/humans/brucellosis-in-dogs/guidelines/brucellosis-in-dogs-vets

Infected dogs are a potential source of infection for people. The illness in people can be severe, protracted and potentially fatal. Infected dogs pose a risk to people through contact with urine, saliva and reproductive materials of infected dogs.

All people involved in sample collection, handling of the dog and clean up, must wear **minimum PPE** of:

- Eye protection
- P2 mask
- Disposable overalls
- Disposable gloves
- Enclosed footwear

and use good personal hygiene.

Pregnant women are vulnerable to severe disease, and should avoid all contact with infected dogs, other dogs that have been in contact with the infected dog.

Disinfectants which inactivate *Brucella* spp. on contaminated surfaces include 2.5% sodium hypochlorite (bleach) and 2-3% caustic soda. Contaminated skin may be cleaned with ethanol, isopropanol, iodophores (Iodine), substituted phenols or dilute hypochlorite. Direct sunlight and autoclaving inactivate Brucella spp.

Sample Collection and Testing: Dogs with suspect clinical signs and the presence of risk factors should be tested for *B suis*. Tissue samples must be submitted fresh, double bagged/potted (with at least 1 hard container), with clear identification that the package contains 'Brucella Exclusion Tissue' (e.g. a piece of paper immediately visible when package is opened). Whole blood or a serum sample is acceptable for serology. At least 1 ml of serum is required.

***Brucella suis* is a notifiable disease:** Under Queensland legislation, if you suspect the presence of this disease, you must report it to Biosecurity Queensland.

Call Queensland Government on **13 25 23** or Emergency Disease Watch Hotline **1800 675 888.**

Also refer to Y:\SVS\SVS-OHS-Public\VTH\SMALL ANIMALS HOSPITAL\Brucellosis guidelines

10.Disinfectants

Virkon S

Purpose

To inform all UQ VTH staff of appropriate use and handling of Virkon S

Procedure

Virkon S completely deactivates Parvovirus, Cat Flu, Ringworm spores and hyphae and Canine Cough in one minute at dilution 1:50.

Virkon is a highly water soluble pink powder with a built in pink colour potency indicator whereby solution changes colour from pink to clear as loses potency. Once prepared Virkon is 100% stable for approximately 7 days.

Virkon S consists mainly of inorganic salts which decompose to harmless by-products – thus is considered environmentally friendly.

Virkon S has a high safety profile towards users / animals and has no significant toxicity implications. Note if a small amount of spray should land on an animal, this will be perfectly safe - avoid eyes.

WHEN TO USE:-

After any infectious/ potentially infectious animals have been in the UQ SAC&VTH.

Pre-cleaning by removing any organic material is required. Please ensure any organic material bagged, sealed and disposed of.

HOW TO PREPARE:-

High Risk situations use dilution 1:50

To prepare a 1% Virkon solution ie 1:100 add 10 gm (one scoopful) per litre of water. Adding Virkon to water not water to Virkon.

To prepare a 2% Virkon solution ie 1:50 add 20 gm (two scoopful) per litre.

HOW TO USE:-

Virkon should be sprayed on a semi-dry, pre-cleaned surface.

It is not necessary to wash Virkon off surfaces following application. It is preferred that the solution is allowed to dry before reusing surface. Some residue may persist but this can be removed later with fresh water.

- Remove solid waste prior to disinfecting
- Clean walls to a height of 1.5 metres
- Metal surfaces should be wiped dry after 15 minutes
- Always wear gloves and other PPE as described in the MSDS when handling disinfectant concentrate.
- Replace lids after use
- Disinfectants should be added to water - not water to disinfectant

WHS ISSUES:-

Virkon is classified as non-irritant to eyes and skin at 1% in-use dilution. There are no occupational exposure limits. The solution is non-irritant to skin and eyes and does not have a harmful vapour phase.

F10 veterinary Disinfectant

Instructions for use:

For hard surfaced and equipment:

- Clear away debris
- Clean and pre-rinse with water
- Apply diluted 1:250 in water as a wash, leave for 20 minutes, wash with water and leave to air dry.

Safety Datasheet for F10 is
http://www.vetnpetdirect.com.au/core/media/media.nl/id.197681/c.1032112/.f?h=3f696eb050e2891de2ae

11.Related Documents

UQ Veterinary Teaching Hospital & Small Animal Clinic – Administrative Procedures: Kennel Cough and Infectious Diseases

UQ Veterinary Teaching Hospital & Small Animal Clinic – Administrative Procedures: Disinfectants – Virkon S.

School of Veterinary Science. Biosecurity and Infection Control Procedures. Large Animal Ambulatory Practices.

School of Veterinary Science. Biosecurity and Infection Control Procedures. Equine Hospitals and Clinics.

CLEANING & DISINFECTION POSTER

CLEANING & DISINFECTION ARE AN IMPORTANT PROCESS IN BIOSECURITY & INFECTION



Cleaning and Disinfection Principles

Cleaning involves the removal of visible debris from surfaces with soap or detergent

Disinfection involves the application of a chemical in order to kill microbes that cannot be removed by cleaning

DISPOSABLE GLOVES SHOULD BE WORN WHEN CLEANING AND DISINFECTING AND PROPER HAND HYGEIENE SHOULD BE CARRIED OUT AFTER ANY CLEANING ACTIVITY

Cleaning and Disinfection Procedures

Ensure all areas are well ventilated during cleaning & disinfecting and disposable gloves are worn

First clean the surface by removing dried-on or sticky debris from surfaces using a cloth/paper towel and a kennel disinfectant (use correct dilution rates).

Follow cleaning using a higher grade disinfectant (Virkon S*).

Floor surfaces should be mopped using either a stable/kennel disinfectant or a higher grade disinfectant (Virkon S*). Replace mop heads regularly.

Always apply cleaners and disinfectants according to the product label, paying particular attention to dilution rates and required contact time

*Refer to SOP on Disinfecting

HAND HYGIENE POSTER

HAND HYGEINE IS THE MOST IMPORTANT WAY OF PREVENTING INFECTIONS IN THE CLINICAL SETTING



Effective hand hygiene removes microorganisms on the skin while maintaining hand health.

Antibacterial soaps should be used throughout the hospital, particularly in high risk areas such as the ICU.

There are multiple hand washing stations situated throughout the Veterinary Teaching Hospital.
These stations use Microshield 4 (Chlorhexidine Gluconate 4%) in disposable pump dispensers.
Bar soaps are not acceptable in veterinary practice settings because of the potential for transmission of pathogens from one person to another.

When Hand Hygiene Should Be Performed

- After removing gloves
- After contact with body substances
- Before and after each patient
- Before eating, drinking or smoking
- After going to the toilet

How?

1. Wet hands with running water
2. Place soap in palms
3. Rub hands together to make a lather
4. Wash hands vigorously for 20 seconds and rinse under running water
5. Dry hands with a disposable towel
6. Turn off tap using the disposable towel

Using hand rubs:

1. Place alcohol-based hand rub in palms
2. Apply to all surfaces of hands
3. Rub hands together until dry

ISOLATION POSTER

ISOLATION ROOMS ARE USED TO HOUSE PATIENTS WITH POTENTIALLY CONTAGIOUS CONDITIONS

Isolation Principles

The isolation room is a designated area to care for and house patients with potentially infectious conditions such as Canine Cough, Parvo or Cat Flu.

The isolation area must be in a low traffic area.

Equipment and materials within the isolation area must be designated for isolation use only, and should not be removed from isolation without being disposed of or disinfected.

ACCESS TO ISOLATION SHOULD BE LIMITED TO THE MINIMUM NUMBER OF ESSENTIAL PERSONNEL NECESSARY TO PROVIDE THE APPROPRIATE CARE

Isolation Procedures

All personnel entering the isolation area must wear the appropriate PPE (ie. Disposable gowns, disposable shoe covers, disposable gloves).

Upon exiting the isolation area, all disposable items worn must be discarded in the clinical waste bin which can be found by the exit door. Reusable gowns or coats used in isolation must be washed after each use.

Disposable shoe covers must be worn and removed upon exiting.

Animals that are housed in isolation are not to be walked, or be allowed to urinate or defecate in areas used by other animals.

If an infectious patient requires procedures within the clinic, it must be transported on a trolley to avoid contamination of the clinic floor. These procedures should also be performed at a time where there is the least amount of patient and personnel traffic within the clinic (ie. last procedure of the day) and these areas must be disinfected according to disinfecting protocols following the procedure/s.

PERSONAL PROTECTIVE EQUIPMENT (PPE) POSTER

PERSONAL PROTECTIVE EQUIPMENT IS AN IMPORTANT ROUTINE INFECTION CONTROL TOOL



Also refer to http://www.ava.com.au/sites/default/files/AVA_website/pdfs/Resource%207%20-%20PPE%20for%20dental%20and%20obstetric%20procedures.pdf for PPE for dental and obstetric procedures.

Personal Protective Equipment Principles:

PPE use reduces the risk of contamination of personal clothing and reduce transmission of pathogens between patients by veterinary personnel.

Protective outerwear such as lab coat or scrubs must always be worn whenever personnel are working in the clinical environment.

PPE USE REDUCES THE RISK OF CONTAMINATION AND TRANSMISSION OF PATHOGENS BETWEEN PATIENTS, STUDENTS AND VETERINARY PERSONEL

Personal Protective Equipment Procedures:

The following PPE must be utilised when appropriate within the clinic:

- Lab coats and overalls
- Scrubs
- Respiratory protection
- Facial/eyes protection
- Gloves
- Footwear

The handrubbing technique for surgical hand preparation must be performed on perfectly clean, dry hands.
On arrival in the operating theatre and after having donned theatre clothing (cap/hat/bonnet and mask), hands must be washed with soap and water.
After the operation when removing gloves, hands must be rubbed with an alcohol-based formulation or washed with soap and water if any residual talc or biological fluids are present (e.g. the glove is punctured).

Surgical procedures may be carried out one after the other without the need for handwashing, provided that the handrubbing technique for surgical hand preparation is followed (Images 1 to 17).

1

Put approximately 5ml (3 doses) of alcohol-based handrub in the palm of your left hand, using the elbow of your other arm to operate the dispenser

2

Dip the fingertips of your right hand in the handrub to decontaminate under the nails (5 seconds)

3

Images 3–7: Smear the handrub on the right forearm up to the elbow. Ensure that the whole skin area is covered by using circular movements around the forearm until the handrub has fully evaporated (10-15 seconds)

4

See legend for Image 3

5

See legend for Image 3

6

See legend for Image 3

7

See legend for Image 3

8

Put approximately 5ml (3 doses) of alcohol-based handrub in the palm of your right hand, using the elbow of your other arm to operate the dispenser

9

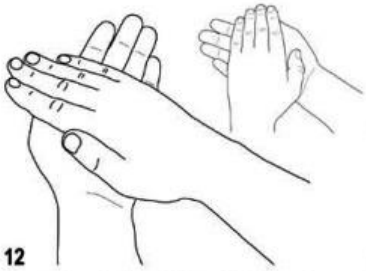
Dip the fingertips of your left hand in the handrub to decontaminate under the nails (5 seconds)

10

Smear the handrub on the left forearm up to the elbow. Ensure that the whole skin area is covered by using circular movements around the forearm until the handrub has fully evaporated (10-15 seconds)

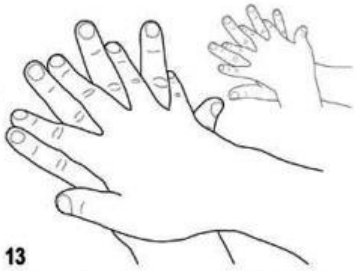
11

Put approximately 5ml (3 doses) of alcohol-based handrub in the palm of your left hand, using the elbow of your other arm to operate the distributor. Rub both hands at the same time up to the wrists, and ensure that all the steps re-presented in Images 12-17 are followed (20-30 seconds)



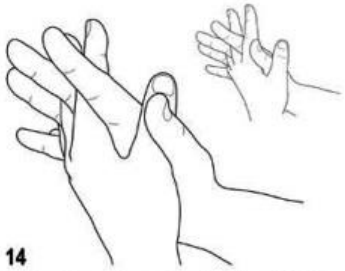
12

Cover the whole surface of the hands up to the wrist with alcohol-based han-drub, rubbing palm against palm with a rotating movement



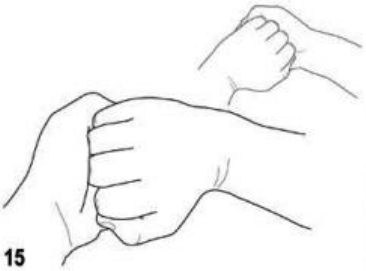
13

Rub the back of the left hand, including the wrist, moving the right palm back and forth, and vice-versa



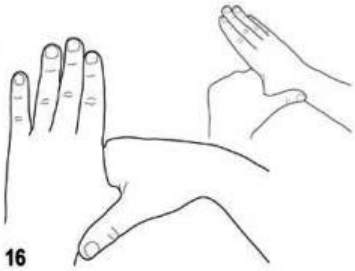
14

Rub palm against palm back and forth with fingers interlinked



15

Rub the back of the fingers by holding them in the palm of the other hand with a sideways back and forth movement



16

Rub the thumb of the left hand by rota-ting it in the clasped palm of the right hand and vice versa

17

When the hands are dry, sterile surgical clothing and gloves can be donned

Repeat the above-illustrated sequence (average duration, 60 sec) according to the number of times corresponding to the total duration recommended by the manufacturer for surgical hand preparation with an alcohol-based handrub.

These have been adapted from WHO Guidelines on Hand Hygiene in Health Care: First Global Patient Safety Challenge Clean Care Is Safer Care. 2009 The full article can be accessed at https://www.ncbi.nlm.nih.gov/books/NBK144036/