ISSN-2220-1181

SAPIENTIA ET ARTE CUSTODIMUS

SOUTH AFRICAN SOCIETY
OF ANAESTHESIOLOGISTS (SASA)

Guidelines for infection control and prevention in anaesthesia in South Africa

2021

Contents

South Afr J Anaesth Analg • 2021 • Volume 27 • Number 4 • Supplement 1

Southern African Journal of Anaesthesia and Analgesia. 2021;27(4 Suppl 1):S1-55
https://doi.org/10.36303/SAJAA.2021.27.4.S1

South Afr J Anaesth Analg
ISSN 2220-1181 EISSN 2220-1173

GUIDELINE

Guidelines for infection control and prevention in anaesthesia in South Africa

This is a consensus document produced by members of a working party established by the South African Society of Anaesthesiologists.

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Chapter 1: Introduction

"The very first requirement in a hospital is that it should do the sick no harm." – Florence Nightingale

Primum non nocere (first do no harm), the ancient adage inferred in the *Hippocratic oath* is a potent reminder of the risk and potential harm associated with the practice of medicine.[1] Hospital-acquired infections (HAIs) cause significant morbidity and mortality to patients and deplete already constrained healthcare budgets. It is estimated that approximately one in seven patients entering hospitals in South Africa is at high risk of acquiring HAIs.[2] The irony of unsafe infection control practices is that they may place patients at risk of greater morbidity or mortality than would derive from the illness being treated. Appropriate anaesthesia practices can decrease the incidence of HAIs.[3]

Poor infection control practices in anaesthesia in South Africa have been lamented in previous studies. The reinforcement of the basic tenets of infection control has been called for, as well as the need for a national guideline to prevent HAIs.[4-7] Health care in South Africa encounters different challenges to those of many other countries. In addition, different regions face unique challenges in South Africa. The high prevalence of human immunodeficiency virus (HIV) in South Africa (up to 65% in seroprevalence studies in certain areas, coupled with the high prevalence of hepatitis B in adults of 8.3–10%),[8] creates an optimal environment for the possible transmission of blood-borne viruses following poor infection control in hospitals. Also, higher rates of HIV and hepatitis B co-infection have been noted in rural areas where adequate infection control may be more difficult to achieve, i.e. 6% (urban) compared to 16.2% (rural).

Thus, the high prevalence of infectious diseases and tuberculosis emphasises the need for evidence-based and strict infection control policies. Furthermore, the narrow margin for error in infection control in South African hospitals is reiterated. Therefore, health policy needs to balance cost constraints with the economic burden of HAIs.

Where there is harm, mention of the law often follows. In keeping with international legal trends in medical liability and litigation,[9] the decision as to whether or not a hospital will be held legally liable for harm to patients as a result of HAIs is dependent on the following:

- Has the hospital introduced best practice infection control measures?
- Has the hospital negligently or intentionally failed to implement designated infection control measures?
- Have hospital staff members, while acting in the course and scope of their employment, negligently or intentionally failed to comply with hospital infection control measures and caused harm to patients?[10]

While economic benefit and legal concerns are important reasons to promote infection control practices, non-maleficence must be

the overriding principle of all policy and decision-making in this regard. Therefore, it is the heart of these guidelines.

With the current SARS-CoV-2 (COVID-19) pandemic wreaking havoc across the globe, protection of healthcare workers and their patients has been pushed to the forefront. Healthcare workers must first ensure that they are optimally protected against infections so that they do not transmit pathogens to their patients and that they are healthy enough to provide medical services to patients. The pandemic forced healthcare workers to strongly consider infection prevention and control strategies. With a receptive audience, this is probably the ideal time for the release of this updated version of the infection control guidelines.

Two new chapters have been added to this version. One is aimed at environmental considerations with regards to infection control whilst the other one deals with basic measures when caring for the already infectious patient.

Chapter 2: Summary of content

General principles of infection control

General principles

- A senior member of the anaesthesia staff should be appointed at each hospital to liaise with the infection control team to ensure compliance with best practice standards in infection control in all areas of anaesthetic practice.
- There must be regular training of healthcare workers in infection control practices. This training must be coupled with monitoring and regular auditing of infection control practice.
- Teaching and training programmes in the practice of anaesthesia should integrate and promote infection control practices as a fundamental part of the curriculum and the speciality.
- The manufacturer's recommendations should always be consulted to determine the compatibility of the respective piece of equipment with decontamination procedures and disinfectants.
- Changing to single-use anaesthesia devices is the best choice in the prevention of cross-infection. However, care must be taken when choosing a single-use device for an institution. There is a wide range of cheap, disposable anaesthesia devices, some of inferior quality. Certainty should always be established as to whether or not the chosen device is fit for purpose and is an evidence-based choice.

Chain of infection

How microorganisms spread

The chain of infection consists of six links and the healthcare worker's role (among others) is to prevent or stop the spread of infections by breaking the chain.

Causative agent

Examples of causative agents are bacteria, viruses, fungi, and protozoa. They are very common in our environment, and most of them are harmless or even essential for the human being. Patients are usually more likely to get an infection from virulent microorganisms due to underlying illness and/or other factors that have weakened the patient's immune system.

Reservoir

The second link in the chain is the reservoir, the place where the causative agents live and multiply. Humans are the most important reservoirs for HAIs; the nose may harbour bacteria and viruses (the MRSA-bacteria is often found here), the skin can carry fungi, and the gastrointestinal tract is a reservoir for many different types of organisms such as viruses, bacteria, bacterial spores, and parasites. The host can be asymptomatic and still transmit pathogens. There are also environmental reservoirs such as soiled linen, dirty gloves, environmental surfaces or improperly cleaned surgical instruments.

Portal of exit from the reservoir

The portal of exit is where the causative agent gets out of the reservoir. Body fluids and tissue from various body systems are common sources of infections.

Modes of transmission (See Table I)

In order for an organism to get from one person to another or from one place in the body to another, it must have a mechanism of getting there known as the mode of transmission.

A lot of effort is aimed at preventing the spread of microorganisms from the reservoir to the susceptible host. The single most important factor for preventing the spread of infection is compliance to hand hygiene strategies.

Portals of entry

The portal of entry is often the same as the portal of exit; this is where the causative agent enters the new host. Medical procedures such as IV catheters, surgical wounds, and intubation are examples that are present in the perioperative period.

Susceptible host – The patient

The final link in the chain of infection is the susceptible host. As mentioned earlier, some bacteria are harmless unless the immune system is weakened. Other factors that can change the outcome could be the presence of invasive devices such as catheters or the absence of natural barriers such as open wounds and burns.

Table I: Modes of transmission

Direct contact	
Physical contact with patient and body fluid, soil or vegetation, e.g. sexually transmitted diseases, blood-borne infections or direct contact with infected wounds	
Droplet spread	• Airway instrumentation such as intubation, suctioning and extubation • Sneezing, coughing, singing • Large particles > 5 µm • Due to the size of the particle, it usually only travels for a short distance usually less that one meter
Indirect contact	
Contact with contaminated environment, linen, hands, etc.	
Airborne/ aerosol spread	• Airway instrumentation such as intubation, suctioning and extubation • Particles < 5 µm • Remains suspended in air for prolonged periods • Spread over large area and possibly further than physical barriers such as rooms or operating theatres • Can be deposited on environmental surfaces
Vehicle spread	• Contaminated hands • Inanimate objects such as laryngoscopes, pens, cell phones
Vector spread	• Flies, mosquitoes

Safe injection practices and preventing the contamination of medication and fluids

Needles and syringes

- Needles and syringes are sterile items, intended for single-patient use only.
- A syringe and needle should be considered to be contaminated after contact with a patient, infusion bag or administration set, and must only be used for that patient. These include syringes used in infusion pumps.
- Medication should not be administered to different patients from the same syringe, even if a new sterile needle is used for each patient. Changing the needle, but not the syringe, is unacceptable practice.
- A syringe must not be reused, or a used syringe reinserted into a medication vial or solution bag or container, e.g. a saline, flush or phenylephrine bag, even if it is for use in the same patient.
- A used needle must not be reinserted into a multiple-dose vial or solution bag or container, e.g. a saline, flush or phenylephrine bag, even if it is for use on the same patient.
- The presence of a non-return valve (one-way valve) or the use of a syringe driver or infusion pump does not permit the reuse of syringes or their contents as it does not prevent the risk of blood contamination. The presence of a check valve (one-way valve or non-return valve) in the infusion set does not prevent blood contamination of syringes or needles.
- Before use, prepared syringes should be capped to avoid contamination.
- After use or at the end of the anaesthetic used syringes and needles should be discarded appropriately.
- Syringes must never be stored nor transported in clothing or pockets.

Preservative-free (single-dose) ampoules or vials

- Preservative-free (single-dose) ampoules or vials are single-dose, single-patient items.
- Do not give drugs from preservative-free vials or ampoules to several patients or save the remaining contents for later use.
- Use of single-dose vials is preferred whenever possible over the use of multi-dose vials for parenteral medications.
- Single-dose vials must be disposed of after the drug dose has been drawn up, and not reused for other patients.
- Cleanse the vial's rubber septum before entering, or the neck of glass ampoules before breaking, with an alcohol swab. Allow drying before entering or breaking the vial or ampoule.

Multi-dose vials

- Use of single-dose vials is preferred whenever possible over that of multi-dose vials for parenteral medications.
- If multi-dose vials must be used, then cleanse the vial's rubber septum with an alcohol swab and allow it to dry before entering the vial. Even vials used for the first time should be cleaned as the cover does not guarantee sterility.
- A new sterile needle and syringe must be used each time the vial is entered.
- Discard a vial if there is suspicion that sterility has been compromised.
- Never leave a needle, cannula or spike device (even if it has a one-way valve) inserted into a medication vial rubber stopper because it leaves the vial vulnerable to contamination.

Infusions, administration sets or items in contact with the vascular system or other sterile body compartments

- These are for single-patient use. They should be discarded after use.
- Bags or bottles containing intravenous (IV) solution should never be used as a common source of supply for more than one patient, e.g. phenylephrine solutions and saline bags for flushing. The time between spiking the vacoliter and administration to the patient should be minimised.
- Never use cannulae or spiking devices, even with a non-return valve, to remove fluid from infusion bottles or bags for several uses or patients.
- Use single-dose, single-use containers for flush solutions.
- Aseptic techniques should be used when preparing infusions and breaks or taps in the lines kept to a minimum.
- Always clean IV injection ports or stopcocks with alcohol and allow to dry before use.
- IV cannula caps are not to be collected for reuse on other patients. These are single-patient, single-use items.
- Both the syringe and the needle or cannula must be sterile when any medication vial or solution is accessed.

Non-injectable items

- Examples include topical drugs, ointments and lubricating gels.
- Ideally, these items should be single-patient, single-use.
- If single-use practice is not possible, utmost care should be taken to avoid self-contamination of these items.
- If contamination is suspected or confirmed, the item should immediately be discarded.

Protecting clean supplies in the anaesthetic cart

- The anaesthetic cart should have its accessible outer surfaces wiped clean in between cases.
- Always perform hand hygiene before the drawers are opened and contents handled.
- The inside of the anaesthetic cart should be cleaned periodically.

Expiration time for drugs and intravenous solutions that are prepared by the anaesthetist

- Provider-prepared drug solutions should ideally be used within one hour of preparation.
- There is paucity on data to determine how long drawn-up drugs or solutions can be used. In the absence of clear direction, provider-prepared drugs can be used to the end of the case. Drugs or solutions should be discarded at the end of the case, whether they were used or not.
- Propofol should be discarded after six hours of ampoule opening. For continuous IV infusions in the ICU, both the tubing, the container and any unused propofol must be discarded after 6–12 hours, as per the manufacturer's recommendations, or when the container is changed.

Hand hygiene guidelines

- Hand washing is one of the most effective infection control practices.
- Gloves do not fully protect against contamination.
- Indications for hand hygiene:
  - Before and after direct patient contact.
  - Before putting on sterile gloves.
  - Contact with body fluids, mucous membranes, open skin and wound dressings.
  - Before making contact with a clean site after touching a contaminated site.
  - After touching a high-touch environmental surface/equipment near the patient.
  - After removing gloves.
  - Before eating or drinking.
  - After using the bathroom.
- Plain soap (non-antimicrobial): Used for routine handwashing when hands look dirty. It will only remove loosely adherent transient bacteria through mechanical friction. It will not decontaminate the hands.
- Antimicrobial soap: Used if there has been visible hand contamination with blood or body fluids. Chlorhexidine has adequate cover against Gram-positive, Gram-negative organisms and viruses but poor cover against mycobacterium and fungi. Iodine compounds have a slightly decreased action against fungi and good cover against all the other microorganism groups. Both chlorhexidine and iodine compounds have an intermediate onset of action and thus need adequate contact time. Chlorhexidine retains its efficacy in the presence of blood and has a lower incidence of adverse skin reactions.
- Alcohol-based hand rubs can be used provided the hands are not obviously dirty or contaminated with proteinaceous material. If an alcohol hand rub is used, it is important to keep the hands and forearms wet during the whole procedure. Using inadequate volumes of alcohol-based rub (0.2–0.5 ml) is as efficient as washing with plain soap and water. Approximately 15 ml of alcohol-based rub is required. Roughly one minute should be spent rubbing the forearm. Thereafter, hands and fingers should be rubbed in the same manner as for handwashing. Alcohol has a good cover against Gram-positive and Gram-negative organisms and also against mycobacteria and viruses. It also has a fast onset of action and should be allowed to evaporate. It has virtually no activity against spores and protozoal oozytes. Physical handwashing with antimicrobial soap and vigorous washing and rinsing is important for spore-forming organisms, such as *Clostridium difficile* and *Bacillus anthracis*.
- Nails: Artificial nails and nail polish should not be used in the operating rooms or ICU. Fingernails should be kept short and clean. Avoid using nail brushes during hand washing as they damage the skin and therefore increase the risk of contamination by microorganisms.
- Non-sterile gloves should be worn whenever contact with blood, body fluid, mucous membranes, non-intact skin and potentially infectious materials is anticipated. They need to be removed as soon as possible and therefore be changed between different procedures on the same patient. They should not be reused. Gloves must be removed before touching equipment if they were in contact with the patient. Curtains, clinical notes, pens, computer keyboards, and cellular and landline telephones must not be touched with contaminated gloves.
- Alcohol-based hand rubs used on gloved hands can be considered in settings with inadequate supply of gloves to meet the above requirements. This practice may potentially degrade the quality of the gloves, making them prone to leakage.
- Bare below the elbow: Different societies have conflicting recommendations with regards to coverage of the arms. This contentious issue has led to multiple guideline changes. Unfortunately, studies have had conflicting results. The proponents of "bare below the elbow" argue that garments have a high risk of being contaminated and by removing them as far away as possible from the hands will decrease the risk of contamination of the hands and increase the efficacy of

handwashing. Opponents to "bare below the elbow" argue that the skin sheds millions of squamous cells every day that contain both normal skin flora and pathogenic microorganisms. By covering up the arms to the wrists, the load of squamous cells entering the environment will be decreased. False nails, nail polish, wristwatches and stoned rings should not be worn.

Anaesthetic equipment decontamination

Management of reusable equipment:

- Reprocessing refers to infection control procedures for removing and inactivating microorganisms on reusable patient-care equipment.
- Reprocessing of reusable patient-care equipment includes cleaning, disinfection, and sterilisation.
- The choice of reprocessing method must consider the instument manufacturer's recommendations.
- Factors that need to be considered are compatibility among equipment components and materials, chemicals to be used, heat and pressure tolerance of the equipment, and time and temperature requirements of the reprocessing methods.

Spaulding classification

Spaulding developed a classification of instruments into "critical", "semi-critical" and "non-critical". This classification is more than 50 years old and is used to determine the level of disinfection for equipment items.

Critical instruments

- Critical instruments have a high risk of contamination.
- These include those instruments that are in contact with sterile sites within the body or the intravascular system.
- Critical instruments should be subjected to sterilisation such as pressurised steam or low temperature methods such as ethylene oxide gas.

Table II: Definitions and classifications used in infection control practices*

Decontamination	A process of removing pathogenic microorganisms from an object or surface so that it is no longer capable of transmitting infectious particles. It is a combination of the processes of cleaning, disinfection, and/or sterilisation.
Cleaning	Cleaning is the removal of foreign material (e.g., soil, organic or inorganic material) from objects and is normally accomplished using water with detergents or enzymatic products (depending on the type of instrument/device/surface that needs to be "cleaned"; surface will be a chemical and instrument will be an enzymatic detergent). **Thorough cleaning is required before** high-level disinfection and sterilisation because inorganic and organic materials that remain on the surfaces of instruments/devices interfere with the effectiveness of these processes. Cleaning is considered the most important step in the reprocessing process of an instrument/device or equipment.
Disinfection	The disinfection process reduces the number of pathogenic microorganisms on the instruments significantly by removing and/or killing them. Bacterial spores are not necessarily killed by disinfection; however, the number may be reduced as a result of the cleaning process. Disinfection may involve chemical or thermal means. **Chemical disinfection** Chemical disinfection is used for equipment that cannot be disinfected in the washer-disinfector and it is used for skin and surfaces. Microorganisms present a range of resistance to chemical disinfectants and no single disinfectant is effective in all situations. Examples of chemical disinfectants are chlorine compounds, alcohol, iodine vapour, and chlorhexidine. **Thermal disinfection** Thermal disinfection of instruments/equipment is achieved with moist heat, either with water or steam. To determine which temperature to disinfect in and for how long, the standard ISO 15883 for washer-disinfectors has specified a so-called A0-value. A0 value explains the relationship between time and temperature, in short, high temperature = less time, and low temperature = more time. **High-level disinfection (HLD)** Destroys all microorganisms (mycobacteria, vegetative bacteria, viruses and fungal spores), except large numbers of bacterial spores in a relatively short exposure time. *Examples of disinfectants:* Glutaraldehyde, ortho-phthalaldehyde, hydrogen peroxide and peracetic acid. Used for semi-critical instrument decontamination. **Intermediate-level disinfection (ILD)** Destroys mycobacteria, vegetative bacteria, most viruses, and most fungi, but does not kill bacterial spores. *Examples of disinfectants:* 70% isopropyl alcohol, iodophor and phenolic compounds, concentrated quaternary ammonium compounds, e.g. hospital cleaners and disinfectants with a tuberculocidal claim. Used for non-critical instruments and environmental surfaces when a tuberculocidal agent is necessary. **Low-level disinfection (LLD)** Destroys lipid or medium-sized viruses, some fungal spores and vegetative bacteria. *Examples of disinfectants:* Diluted quaternary ammonium compounds, e.g. hospital cleaners and disinfectants without a tuberculocidal claim. Used for non-critical items and surfaces when a tuberculocidal agent is not needed.
Sterilisation	A process whereby all types of microorganisms, e.g. mycobacteria, vegetative bacteria, viruses and fungal spores, including bacterial endospores, are eliminated. To be effective, sterilisation must be preceded by cleaning, the removal of all foreign material from the item, and disinfection, the reduction of pathogen microorganisms to a level that is not harmful to health. *Examples of methods* include pressurised steam (autoclaves) or low-temperature sterilisation methods, e.g. ethylene oxide gas and hydrogen peroxide plasma, as well as hot air ovens. It is used for critical instrument decontamination.

*Reproduced with permission[2]

- Examples include surgical instruments, implants and sterile catheters.
- With the advent of multidrug-resistant and aldehyde-resistant organisms there was a call to also include other semi-critical instruments into the category of critical instruments. One such an item is endoscopes. The design is flawed with lots of crevices that may retain microorganisms. A study by Ofstead showed that almost 50% of endoscopes retained moisture that can aid in the development of a biofilm. In addition, 71% of endoscopes showed microbial growth and cultured organisms such as *Stenotrophomonas maltophilia* and *Citrobacter freundii*.
- Other items that are relevant to the anaesthetist include airway instruments such as laryngoscope blades and handles as well as forceps. Airway instrumentation is often associated with trauma to the mucosa. Several studies have demonstrated occult blood on these items. These instruments thus meet the criteria for critical instruments. These items require cleaning and disinfection followed by sterilisation as required and as per manufacturer's recommendations.

Semi-critical instruments

- Semi-critical instruments have an intermediate risk of contamination.
- These instruments only come into contact with intact mucosa and intact skin.
- Examples given under this category include endoscopes, respiratory equipment and cystoscopes. It is, however, interesting to note that most of these items are now being considered "critical instruments".

Non-critical instruments

- Non-critical equipment has a low risk of contamination.
- These instruments only come into contact with intact skin.
- Divided into patient non-critical items such as blood pressure cuffs and oximetry probes and environmental non-critical items such as cot sides, theatre trolleys and uncontaminated floors.
- Cleaning and drying are usually adequate.

Laryngoscopes

Laryngoscope blades

Contaminated anaesthetic equipment has been implicated in the nosocomial transmission of infectious diseases. Options for the reprocessing of laryngoscope blades include:

- Use of disposable (single-use) laryngoscope blades (DLBs) (preferred): The metal type only must be used. They should be discarded after single use. DLBs should not be reused, even after sterilisation.
- The sterilisation of reusable laryngoscope blades (RLBs): The light intensity of all RLBs that are steam sterilised should be monitored. Handling and storage are important.
- HLD: There are significant concerns about the use of HLD in a South African setting when decontaminating RLBs. Evidence of poor compliance with HLD protocol has been documented, and there is a significant margin for human error in the HLD protocol. If it is used, hospitals must have a specific step-by-step instructional protocol in print that is well understood. Decontamination should occur in a specific designated area, away from patients and other healthcare workers. Frequent in-service training of anaesthesia nurses on HLD must be conducted. The decontamination of RLBs should be monitored and audited for compliance.
- No other method of disinfection, e.g. chlorhexidine and alcohol, should be used to decontaminate RLBs.

Laryngoscope handles

The laryngoscope handle should be decontaminated after each patient. To minimise laryngoscope handle contamination:

- Remove the blade from the handle immediately after use and place the contaminated blade in a receptacle.
- Do not close the contaminated blade on the handle after intubation.
- Consider covering the handle with a new disposable plastic bag for each patient, as described in the rationale.
- Decontaminate by sterilisation, or HLD or ILD.

Sterilisation

Sterilisation steps are as follows:

- Send the laryngoscope handle to the central sterile supplies department for sterilisation.
- Batteries must be removed in the operating theatre (OT).
- Adequate numbers of handles per OT should be acquired to allow for this.
- The manufacturer should be consulted to determine compatibility with the type of sterilisation. Steam sterilisation is recommended. If the handle is not sterilisation friendly, it should be replaced.

High-level disinfection

HLD steps are as follows:

- HLD should take place after each patient.
- Batteries should be removed in the OT prior to HLD.
- There should be a specific step-by-step instructional protocol in print, that can be easily understood.
- HLD must be monitored and audited for compliance.
- Adequate numbers of laryngoscope handles per OT must be acquired to allow for this.

Intermediate-level disinfection

ILD steps are as follows:

- ILD should take place after each patient.
- Chlorhexidine 2% with alcohol 70% should be used.
- HLD or sterilisation must be employed if there is visible blood or organic material contamination.
- Several articles prefer HLD over ILD, so ILD is not the preferred choice.

- After sterilisation or disinfection, the laryngoscope handle and blade should be checked for function and then packaged into a sealed plastic bag to prevent recontamination.

Magill forceps

Magill forceps must be steam sterilised after each use. Adequate numbers of Magill forceps per OT should be acquired to allow for this.

Nasopharyngeal and rectal temperature probes

Nasopharyngeal and rectal temperature probes require sterilisation after each use according to manufacturers' recommendations. Adequate numbers of nasopharyngeal temperature probes per OT should be acquired to accommodate this.

Suction bowl

The suction bowl is the container that is filled with water that is used to clear anaesthetic suction catheters or Yankauers™. It should be changed to a plastic or metal receptacle that can be replaced after each patient (one suction bowl per patient). The contaminated receiver should be sent for sterilisation.

Suction tubing

Disposable plastic tubing is recommended for suction tubing. The tubing should be replaced after each patient.

Oropharyngeal airways

Single-patient use only is applicable to oropharyngeal airway equipment, which must be discarded after each use.

Bougies, and intubation guides and stylets

A gum-elastic bougie may be disinfected up to five times between patients according to the manufacturer's recommendations. It should be stored in a sealed packet.

Alternative single-use intubation aids are preferable to bougie use. Intubation aids and stylets are single-use items. Rigid stylets for use with video laryngoscopes e.g. GlideRite with the Glidescope, should be sterilised as per manufacturer's guidelines.

Breathing filters and breathing circuits

Use of a breathing filter must include the following:

- Use a new, high-quality heat-and-moisture-exchange filter (HMEF) for every patient. The HMEF must be changed between patients.
- The filter should be placed on the Y-piece between the endotracheal or tracheostomy tube and the elbow connector or breathing circuit.
- The high-quality HMEF should be above the level of the lungs, with the filter in a vertical position to decrease the risk of contamination from secretions from the patient or condensate from the breathing circuit.
- The anaesthetist must actively search for complications associated with the use of breathing filters, such as obstruction of the filter with blood or secretions, an increase in airway resistance and possible disconnection.
- The filter should not be placed between the circuit and the absorber as this practice can lead to the desiccation of soda lime, with the resultant risk of carbon monoxide poisoning.
- The filter has to be changed when it becomes visibly contaminated with blood or secretions, or with condensate within the breathing system.

Type of breathing filter

- The HMEF must have been tested using the saline test as prescribed in ISO 9360-1:2000 or the European standard norm EN13328-1. The HMEF should have a 99.97% efficiency at a flow rate of 30 l/minute.
- The HMEF should be able to withstand a pressure of 60 hectopascals (≈ 60 cmH_2O) without allowing liquid to pass through, or 20 hectopascals above the set pressure limit of the breathing circuit.
- The HMEF must have a minimum humidity output of 20 g/m^3 in patients ventilated < 10 hours or 33 g/m^3 in ICU patients ventilated > 10 hours.
- When using low flows, the dead space in the filter should be appropriate for the patient's tidal volume.
- Ideally, the HMEF should be a hydrophobic pleated filter.
- Electrostatic filters should not be used in cases where there is a high risk of cross-infection as they do not prevent the passage of liquid through the filter. Electrostatic filters do not prevent transmission because liquid (carrying viruses and bacteria along) can pass through these filters.
- The increase in dead space, increased airway resistance and possible delayed inhalational induction of anaesthesia when using breathing filters/HMEFs, should be considered in children. The lower-weight limit should be a heat-and-moisture exchanger (HME) of 5 kg and filter of 3 kg.

Breathing circuits

The breathing system consists of the elbow connector or catheter mount, the breathing circuit, the reservoir bag and CO_2 absorber. The components of the breathing circuit can be reused between cases for up to seven days, provided that:

- A high-efficiency filter has been used.
- There are no defects in the system.
- It has been disinfected daily according to the manufacturer's instructions.
- It has been cleared by the manufacturer to be used as such.
- The breathing system components are seen as semi-critical items and should be disinfected according to the manufacturer's instructions.
- The CO_2-absorber canister should be cleaned every time the absorber material is changed. Disinfection must take place according to manufacturer's guidelines.

The components of the breathing circuit should be changed immediately in any of the following circumstances:

- When it is visibly soiled with blood or secretions.
- When used on a patient with a confirmed or potential notifiable infectious disease that involves the risk of transmission via the breathing circuit and reservoir bag, e.g. tuberculosis, acute viral hepatitis, measles, influenza virus, infection and/or colonisation with a multidrug-resistant pathogen and upper or lower respiratory tract infection.

Oxygen tubing, oxygen masks and nasal prongs

- These are single-use items and should be discarded after use on a single patient.
- An area of at least 0.4 m from the mask should be considered to be a potential hazard for aerosolised pathogens.
- Patients with high-risk respiratory infections should only be nebulised when necessary and should be isolated during nebulisation in a room with good ventilation.

Bag valve mask resuscitators

- All resuscitators should be fitted with a high-efficiency breathing filter between the valve and the mask before being used on a patient.
- Resuscitators used on the same patient should be capped at the patient connection port when not in use.
- Resuscitators should be cleaned and disinfected according to the manufacturers' instructions.
- The resuscitator should be disassembled, and all the parts washed thoroughly, using clean water and mild detergent. It is necessary to ensure that the detergent is suitable for the material.
- Do not disassemble the pressure release valve and the positive end-expiratory pressure valve.
- All of the parts should be rinsed in clean water to remove the detergent.
- All of the parts should be allowed to dry in a clean, controlled environment, where the risk for recontamination is low.

The components should then be subjected to one of the following decontamination techniques:

- Pasteurisation for 30 minutes (not the oxygen reservoir bag).
- Autoclaving not to exceed 132 °C (not the oxygen reservoir bag).
- Ethylene oxide gas (all parts are suitable).
- Liquid sterilisation (all parts are suitable) with Cidex OPA® or sodium hypochlorite. Wash thoroughly to remove any excess disinfectant.

Manual resuscitator should be sterilised:

- For first-time use.
- Between patients.
- When visibly contaminated.
- Every 24 hours of use in the same patient.

Supraglottic devices

Single-use (disposable) supraglottic airway devices (SADs) are preferred to reusable SADs.

If reusable SADs, e.g. LMA Classic™, are used, they should be sterilised in an audited sterile service department and not more often than recommended by the manufacturer, e.g. 40 times for LMA Classic™. Do not decontaminate and reuse single-use SADs.

Storage of semi-critical items

Semi-critical items should be packaged and stored in a way that prevents recontamination. Suggested compliant storage methods include a peel pouch or a closed plastic bag. These items should not be left unwrapped in or on top of anaesthesia workstations and trolleys.

Non-critical medical equipment surfaces

Non-critical medical equipment surfaces extend to blood pressure cuffs, stethoscopes, and frequently used control mechanisms, e.g. pop-off knobs, flow controls and vaporisers.

It is necessary to disinfect with a low- or intermediate-level disinfectant after each patient.

Medical equipment surfaces can become contaminated with blood and infectious agents and contribute to the spread of healthcare-associated infection. Loftus et al. reported that multidrug-resistant bacterial transmission to the anaesthesia work area occurred during the practice of general anaesthesia.

Disposable devices

A single-use medical device is to be used on a single patient during one procedure. The reuse of disposable or single-use devices started in the 1970s and is a growing and common practice worldwide, especially in resource-limited settings. Informed consent should be obtained from the patients if an item is to be reused on them. Certain issues exist, including the decontamination technique and the risk of cross-contamination, material alteration, a clear limit to the number of times that an item can be reused, mechanical failure of the device, exposure risks to healthcare workers and ethical and legal implications. Although the reuse of single-use devices is strongly discouraged, the suggested practice from the International Society for Infectious Diseases (ISID) and the World Health Organization (WHO) are as follows:

- A facility should be committed to the reuse of single-use devices and have an institution-specific policy with clear guidelines.
- The disposable devices should be classified and reprocessed as per their intrinsic risk: critical, semi-critical and non-critical.
- Functionality and integrity of the device should be maintained.
- The package labelling and manufacturer's guidelines should be followed.
- Reprocessing of disposable devices should be cost-justified and performed by a licensed reprocessor.
- Both the physician and the patient should be informed that a device being used is a reprocessed single-use device.
- Any person who reuses a single-use device takes full responsibility for its safety and effectiveness.

A platform for reporting any adverse events from the reuse of single-use devices should be available. The Association of Perioperative Registered Nurses (AORN) has recommended that the sterility, integrity and functionality of a reprocessed single-use device must be documented as safe for patient care and/or equal to the original device specifications.

Trans-oesophageal echocardiogram probes (TEE)

A semi-critical device with increasing usage in anaesthetic practice that has the potential for cross-contamination, especially if the probes are damaged. A sheath should preferentially be used but does not eliminate the risk of contamination and does not exclude the probe from HLD. The recommended basic principles for reprocessing TEE probes are:

- Clean the probe shaft and tip either with immersion or with detergent moistened wipe or enzymatic cleaner to remove gross contamination.
- Use a second wipe to wipe the proximal non-immersible parts such as the handles.
- Ensure that there is no structural damage to the probe.
- Use HLD to disinfect the probe tip and flexible shaft.
- Thoroughly rinse and dry before storage.

The manufacturers' instructions regarding chemical disinfectants should be followed.

Prevention of intravascular catheter-related bloodstream infection (CRBSI)

Placement of central venous catheters

- The subclavian site is preferred over either the internal jugular or femoral sites in adult patients in order to reduce the incidence of sepsis and of thrombosis.
- If the patient has chronic kidney disease, consider the internal jugular vein to avoid subclavian vein stenosis.
- In children and in infants there is no preferred venous site to minimise the risk of infection.
- Use ultrasound when possible and when trained operators are available to reduce the time to cannulation, the number of cannulation attempts and the incidence of mechanical complications.
- Use a line with the minimum number of lumens necessary to facilitate management of the patient.
- When adherence to sterile technique cannot be assured, the line must be removed as soon as possible, but within 48 hours.
- An engineered stabilisation device that is designed specifically to control movement at the catheter hub, is recommended, as sutures increase the risk of infection. Standard dressings and tape are not suitable alternatives.
- All lines that are no longer needed should be removed promptly.

Sterile technique for the placement and securing of central venous catheters

- The operator should scrub, as for a surgical procedure, prior to the placement of a central venous catheter.
- Maximal sterile barrier precautions to be used include the use of a cap, mask, sterile gown, sterile gloves and a sterile full body drape.
- For skin decontamination prior to catheter insertion, the application of sterile > 0.5% chlorhexidine gluconate in 70% isopropyl alcohol represents standard of care. If there is a contraindication to the use of chlorhexidine, tincture of iodine, an iodophor or 70% alcohol may be used as alternatives. Caution should be taken to avoid pooling of cleaning solution as this can lead to burns.
- No comparison has been made between using chlorhexidine preparations with alcohol and povidone-iodine in alcohol to prepare clean skin.
- No recommendation can be made for the safety or efficacy of chlorhexidine in infants aged < 2 months.
- The skin antiseptic must be allowed to dry (at least 30 s) prior to performance of the procedure.

Catheter dressing and site management

- Do not use topical antibiotic ointment or creams on insertion sites as they have the potential to promote fungal infections as well as antimicrobial resistance.
- A sterile, transparent semi-permeable polyurethane dressing must be used to cover the site. Sterile gauze and tape may be used as an alternative.
- Sterile gauze should be used if there is any bleeding, exudate or excessive skin moisture that accumulates around the insertion site.
- The dressing must be replaced immediately if there is any sign that it is becoming loose, if there is any soiling, or if any dampness is noted under the dressing or at the insertion site. When replacing the dressing, the skin must be cleaned with antiseptic and allowed to dry before applying the new dressing.
- Gauze dressings must be replaced at least every two days.
- Clear transparent dressings must be replaced at least every seven days.
- Line sites must be monitored daily, especially at the catheter-skin junction site and surrounding area for pain, erythema, swelling or purulence, which may indicate infection, phlebitis, infiltration, or catheter-associated venous thrombosis.

Use of catheters and dressings that have associated antimicrobial activity

- Impregnated sponge-type dressings are only recommended if the central line-associated bloodstream infection (CLABSI) rate is not decreasing despite adherence to basic prevention measures, including education and training. This includes adherence to all of the previously described principles.
- Antimicrobial-impregnated catheters or those with antimicrobial properties may be considered in environments in which the rate of CLABSI is not decreasing, and if lines are likely to remain in place for more than five days.

Management of lines and administration sets

- In patients not receiving blood, blood products or fat emulsions, replace administration sets that are continuously used, including secondary sets and add-on devices, no more frequently than at 96-hour intervals, but at least every seven days.
- Blood product transfusion administration sets and filters should be replaced after the completion of each unit or every four hours. If more than one unit can be infused in four hours, the transfusion set can be used for a four-hour period.
- Administration sets used for intravenous fat emulsions should be replaced every 24 hours or with each new container.
- For continuous IV infusions in the ICU, the tubing, the container and any unused propofol must be discarded after 6–12 hours, as per the manufacturer's recommendations, or when the container is changed.
- When using needleless connectors, use a luer-locking mechanism to ensure a secure connection to the central venous catheter hub or access site.
- When needleless systems are used, a split septum valve may be preferred over some mechanical valves due to increased risk of infection with the mechanical valves.
- Disinfect needleless connectors prior to each entry into the device.
- Use aseptic no-touch technique to change the needleless connector.
- Change the needleless components at least as frequently as the administration set. There is no benefit to changing these more frequently than every 72 hours.
- Needleless connectors and access ports on administration sets must be cleaned with 70% alcohol, tincture of iodine or chlorhexidine, prior to injection or connection.

Placement of peripheral catheters

- Strict hand hygiene must be observed before and after the placement, removal or palpation of the catheter-insertion site.
- Strict hand hygiene is also required before and after accessing or dressing a catheter.
- Clean gloves, rather than sterile gloves, should be used for the insertion of peripheral venous catheters if the access site is not touched after the application of skin antiseptics.
- Sterile gloves should be used for the insertion of arterial and umbilical catheters.
- Clean gloves must be worn when changing the dressings on intravascular catheters.
- Skin preparation with 70% alcohol, tincture of iodine or chlorhexidine is acceptable for peripheral venous catheter insertion.
- The upper extremity should be used for the insertion of venous catheters in adults.
- The upper or lower extremities, or the scalp, may be used as a catheter insertion site in paediatric patients.
- Remove the peripheral catheters if there is any sign of infection or inflammation at the insertion site. This includes redness, tenderness, purulence or obvious thrombophlebitis.

Placement of arterial lines

- The radial or dorsalis pedis site is preferred over the femoral or axillary site to reduce the risk of infection in adults.
- Strict hand hygiene must be observed before and after the placement, removal or palpation of the catheter-insertion site.
- Strict hand hygiene is also required before and after accessing or dressing a catheter.
- Sterile gloves should be used for the insertion of arterial lines.
- Clean gloves must be worn when changing the dressings on intravascular catheters.
- A cleaning solution containing more than 0.5% chlorhexidine in alcohol should be used when inserting arterial catheters.

Infection control recommendations for regional anaesthesia

Central neuraxial techniques

In a patient with known or suspected bacteraemia, prophylactic pre-procedural antibiotic therapy should be considered. Aseptic techniques must be applied during preparation of equipment. A caudal anaesthetic is considered to be a neuraxial technique as the caudal space is a continuation of the epidural space.

Maximal barrier precautions apply:

- Jewellery should be removed and hands washed.
- Caps, masks (covering both mouth and nose), sterile gloves and gowns.
- Sterile drapes.
- Face mask should also be worn by the anaesthetic assistant.
- An antiseptic, preferably a 0.5% solution of chlorhexidine with alcohol, should be used for skin preparation.
- Avoid pooling of the cleaning solution to avoid the risk of burns.
- Allow the solution to dry completely before touching or palpating the back.
- Meticulous care should be taken to avoid contact of chlorhexidine with the cerebrospinal fluid.
- Do not pour chlorhexidine into containers in close proximity to equipment that will be used for the neuraxial anaesthetic. Cover or protect equipment while cleaning with chlorhexidine.
- A sterile occlusive dressing must be applied over the puncture site.
- Bacterial filters may be considered during extended continuous epidural infusion.
- Disconnection and reconnection of the neuraxial delivery system should be limited.
- Consider removing a catheter that has become disconnected without it being noticed as soon as discovered.

Catheters must not remain in situ for longer than is clinically necessary.

Peripheral nerve blocks

- Maximal barrier precautions are generally not necessary.
- Maximum barrier precautions should be used if the patient is immunocompromised or a perineural catheter needs to be inserted.
- Jewellery should be removed and hands washed. Sterile gloves must be worn.
- Aseptic techniques should always be used during the preparation of equipment, e.g. ultrasound, the drawing up of drugs and the placement of needles and catheters.
- An antiseptic, preferably chlorhexidine with alcohol, should be used for skin preparation, and adequate time allowed for drying. Do not allow alcohol to come in contact with an ultrasound probe, needles and catheters.
- Prevent pooling of cleaning solution to avoid burns.

Use of ultrasound

- A sterile probe and handle covering should be used, e.g. a sterile transducer sheath.
- Do not use pre-lubricated condoms as sheaths as the lubricant can damage the probe. (Logiq e manual).
- An antiseptic, preferably chlorhexidine with alcohol, should be used for skin preparation, and adequate time allowed for drying. Do not put the probe on the patient until the alcohol has evaporated to prevent alcohol-induced damage to the probe.
- Do not allow sterilant such as Cidex® to come into contact with the patient as it may cause damage to skin or mucous membranes. If contact does occur, refer to the information leaflet of the specific sterilant.
- The probe should immediately be wiped with a soft towel to remove any gel/lubricant residue and any visible contamination.
- Product information should be consulted as to which cleaning agents are appropriate for the specific machine or probe.
- Use single-use, sterile gel, e.g. a K-Y® lubricating.

Antimicrobial prophylaxis for surgical procedures

Table III: Altemeier classification of surgical wounds

Class I: Clean SSI risk < 1%	Sterile area of body. Skin intact before surgical incision. Surgery does not involve opening of gastrointestinal, respiratory, genito-urinary or oropharyngeal tracts.
Class II: Clean contaminated SSI risk 2–5%	Opening of body cavities: gastrointestinal tract, respiratory tract, genito-urinary tract or oro-pharyngeal tracts in the absence of gross contamination.
Class III: Contaminated SSI risk 5–10%	Massive surgical soiling by gastrointestinal contents or opening of genito-urinary or biliary tracks in patients with tract infections. Recent open traumatic wounds.
Class IV: Dirty SSI risk > 10%	Body site that contains pus, foreign body or faeces. Traumatic open wounds > 4 hours.

SSI – Surgical site infection

There are four considerations to keep in mind when prescribing antibiotic prophylaxis:

1. Who needs antibiotic prophylaxis?
2. What are the factors that influence the antibiotic choice?
3. Timing.
4. Duration of antibiotic prophylaxis.

Procedures that need antibiotic prophylaxis

- Class I surgery involving the placement of prosthesis or implants.
- Class II surgery.
- Class III and IV should be on curative antibiotics.

In procedures where antibiotic prophylaxis would not normally be used, it should be considered in patients with an increased risk for surgical site infections (SSIs) or where infection will be catastrophic. Patients at increased risk for infection include geriatric and oncology patients, those with diabetes, HIV, obesity, transplant patients, etc.

Do not give antibiotic prophylaxis for clean, non-prosthetic uncomplicated surgery.

Choice of prophylactic antibiotics

- Use an antibiotic that is safe, inexpensive, and a bactericidal with an in vitro spectrum that covers the most probable intraoperative contaminants for the operation. Other factors that may influence the choice of antibiotics are, amongst others, renal function, other comorbidities, recent antimicrobial use, institutionalised patients, known colonisation with a drug-resistant organism as well as immunocompetency.
- Use your local antibiotic formulary, and always consider potential adverse effects when giving antibiotics for prophylaxis.
- For a detailed table of surgical procedure and the best choice of prophylactic antibiotics, see Appendix A.

Timing of prophylactic antibiotics

- Give a single dose of antibiotic prophylaxis intravenously 30 minutes before skin incision, but not more than one hour before. Vancomycin should not be used as a first-line prophylactic antibiotic. When it is used, it should be given as an infusion that ends 30 minutes prior to skin incision.
- Give antibiotic prophylaxis earlier for operations in which a tourniquet is used.
- With regards to Caesarean delivery, antibiotic prophylaxis should not be delayed to the clamping of the cord.
- A second dose of an antibiotic with a relatively short half-life, e.g. cephazolin, is often recommended for prolonged procedures or procedures with massive blood loss.

Duration of prophylactic antibiotic therapy

Antibiotic prophylaxis should be brief and limited to the surgical time. It is sometimes used up to 24 hours and, under very exceptional circumstances, up to 48 hours. Prophylaxis should never continue for more than 48 hours.

Environmental considerations in infection control and prevention

Please refer to the relevant IUSS guidelines that can be found at www.iussonline.co.za as well as the SASA website.

Operating theatre ventilation

- Every operating theatre complex requires effective central humidity ventilation and air-conditioning system (HVAC).
- Maintain at least 15–20 air changes per hour of which at least three such air changes should be with fresh air.
- Maintain positive pressure within the operating theatre compared to the corridors and adjacent areas. This is to facilitate the movement of air from the operating theatre along a pressure gradient towards adjacent areas. Air thus moves away from the sterile operating site and reduces the risk of contaminated air getting in contact with the operating field and sterile instruments.
- Ideally, air should come from the ceiling and move towards at least two air exhaust vents located close to the floor.
- Make sure that air exhaust vents are not obstructed or covered.
- All air (re-circulating and fresh air) should pass through filters before entering the operating theatre.
- Keep the doors to the operating theatres closed at all times.
- The air-conditioner system should operate continuously independent of whether there is a case on the table or not.

Forced-air warmers

- Consider using conductive fabric warmers over forced-air warmers for preventing intraoperative hypothermia, especially in high-risk surgery such as joint replacements.
- Where conductive fabric warmers are not available, forced-air warmers should be used as the risk of hypothermia outweighs the possible risk of contamination of the surgical wound.

Operating theatre temperature and humidity

Keep ambient theatre temperature between 18–24 ºC. The ambient temperature should be uniform throughout the space. Keep humidity levels between 30–60%.

Environmental cleaning of the operating theatre

Table IV: Definitions

Cleaning	The physical removal of foreign material, e.g., dust, soil, and organic material such as: blood, secretions, excretions, and microorganisms. Cleaning physically removes rather than kills microorganisms. It is accomplished with water, detergents and mechanical action.
Contact time/ Dwell time	The defined time for which surfaces are exposed to a chemical or thermal disinfection process to achieve the appropriate level of disinfection. Inadequate contact time may lead to incomplete disinfection.
Disinfectant	Product used on inanimate objects to reduce the number of microorganisms to an acceptable level. Hospital-grade disinfectants require a drug identification number (DIN). High-level disinfectants should not be used to clean environmental or inanimate objects.
Disinfection	The inactivation of disease-producing microorganisms with the exception of bacterial spores. Hospital-grade disinfectants are used on inanimate objects. Medical equipment must be cleaned properly before effective disinfection can take place.
Preliminary cleaning	Damp-dust horizontal surfaces prior to first case. Do not clean with dry materials as that causes dust to become airborne. Use a clean, lint-free cloth moistened with low-level disinfectant. Avoid spraying or misting methods. Start at higher surfaces and work down in a clockwise manner. Damp-dust equipment before it is brought into or out of the operating theatre. Inspect operating theatre lights for cleanliness before the first case of the day. Floors should always be considered contaminated even after proper cleaning.
Intraoperative cleaning	The responsibility for verifying disinfection of a contaminated surface rests with the perioperative team member who is first aware of the contamination. All contaminated (blood, body fluids, or other potentially infectious material) items or surfaces occurring intraoperatively are to be promptly cleaned/disinfected as required using facility-approved disinfectant. Equipment leaving the operating theatre is cleaned and disinfected with hospital-approved disinfectant. Chemical spills occurring intraoperatively are to be managed as per site/regional policy/procedure.
Between procedures	Each operating theatre must be cleaned and disinfected immediately after each case. Do not start the process before the patient has left the area. Prior to cleaning, remove all trash, linen, and recycling from the room, including soiled anaesthesia equipment and supplies. All surfaces that have been in direct or indirect contact with the patient or body fluids are considered to be contaminated and therefore are to be cleaned/disinfected with a hospital-approved disinfectant. Contaminated linen should be handled as little as possible.
Terminal cleaning	Cleaning staff should adhere to standard precautions with regards to personal protective equipment (PPE). Staff performing cleaning may be required to wear additional PPE during terminal cleaning after procedures with additional precautions such as the highly infectious patient. Operating theatres are to be terminally cleaned at minimum once every 24 hours during a regular workweek regardless of whether the theatre has been used. All floors should be cleaned using a wet vacuum or single-use mop and a disinfectant (follow dwell time indicated on manufacturer's instructions). Floor cleaning should progress from cleanest area to dirtiest, from perimeter of the room to the centre. Care must be taken to ensure the floor under the theatre bed and trolleys are also cleaned.

- Cleaning of surfaces and instruments with detergents is needed before disinfection and decontamination can take place. The presence of organic matter, salts and obvious contamination can compromise the efficacy of the terminal reprocessing procedures.
- Use Environmental Protection Agency (EPA)-approved disinfectants such as a quaternary ammonium compound to clean all areas that could have been contaminated during the procedure.
- Avoid methods that can lead to mist, aerosols or dispersion of dust.

Surgical attire

Scrubs

- Scrubs should be made from material that is tightly woven, low-linting, stain-resistant and non-flammable.
- It should not be 100% fleece but rather a mixture of cotton with 10–20% polyester.
- Change scrubs that are visibly soiled or contaminated with potential infectious material.
- Reusable scrubs should be washed at a healthcare-accredited laundry.
- For procedures at high risk of contamination from blood or bodily fluids, a waterproof apron should be worn under the surgical gown.
- Surgical scrubs should not be worn outside the operating theatre complex due to the risk of spreading microorganisms.
- Home laundering of scrubs is not recommended as it does not meet specified criteria to reduce microbial load.
- Maintaining good personal hygiene is as important as wearing appropriate theatre attire.

Other theatre attire

- Wear a surgical mask when packs are open and for the duration of the procedure.
- Surgical mask must cover all facial hair, the mouth and the nose.
- Wear a theatre cap that covers all hair on the head and sideburns.
- Shoe covers do not protect against SSIs.
- Shoe covers can be replaced with dedicated theatre shoes that are easily washable and washed at the end of the day.
- Outside shoes should not be worn inside the operating theatre.
- PPE use should be based on the most likely mode of transmission of organisms.

Infection control precautions for the infectious patient

Non-emergency cases should be postponed until the patient is deemed to no longer be infectious. Precautions should be based on the mode of transmission of the specific microorganism.

Modes of transmission

Direct contact

- Physical contact with patient and body fluid, soil or vegetation.
- Droplet spread:
  - Large particles > 5 µm.
  - Short distance spread usually within one meter but can be further as has been demonstrated with smallpox and SARS viruses.
  - Airway instrumentation such as intubation, suctioning and extubation.
  - Sneezing, coughing, singing.

Indirect contact

- Airborne/aerosol spread:
  - Particles are less than 5 µm.
  - Remains suspended in air for prolonged periods.
  - Spread over large area and possibly further than physical barriers such as rooms or operating theatres.
    - Can be deposited on environmental surfaces. Depending on the type of surface, it can remain viable for a couple of hours or up to a couple of days.
  - Airway instrumentation such as intubation, suctioning and extubation.
- Vehicle spread:
  - Contaminated hands.
  - Inanimate objects such as laryngoscopes, pens, cell phones.
- Vector spread:
  - E.g. flies and mosquitoes.

Type of precautions

Standard precautions

- Keep to all the standard precautions discussed in this document.
- Healthcare workers should have documented immunity to hepatitis B virus.

Contact precautions

- Standard precautions as above.
- Adhere to hand hygiene standards.
- Isolation cubicle until no longer infectious.
- Use PPE including a gown.
- Remove PPE before leaving the immediate environment of the patient.
- Take care not to self-contaminate when removing PPE.
- Maintain contact precautions throughout the entire perioperative period.
- Appropriate environmental decontamination of the operating theatre at the end of the case.

Droplet precautions

- Standard precautions as above.
- Adhere to hand hygiene standards.
- Ideally, to be isolated. If isolation not available, keep patient at least one meter from any other patients.

- Educate patients on respiratory hygiene, e.g. coughing and sneezing etiquette.
- Patient to wear a standard face mask when outside the isolation area.
- Healthcare workers must use standard PPE.
- Maintain these precautions throughout the entire perioperative period.

Airborne precautions

- Standard precautions as above.
- Adhere to hand hygiene standards.
- Patient should be in an airborne isolation room.
- Patient should remain in the isolation room except for medical/surgical procedures that require the patient to leave the room.
- Any non-emergency case should be postponed until the patient no longer needs respiratory isolation.
- Patient to wear a standard face mask when outside the isolation area.

Healthcare workers donning of PPE

- Remove jewellery and any other personal items.
- Empty pockets.
- Tie hair back and cover all hair.
- Perform hand hygiene and don disposable apron.
- Perform hand hygiene and change shoes or apply overshoes.
- Perform hand hygiene and don first pair of gloves.
- Hand hygiene and don an impermeable disposable gown or coverall.
  - Ensure that gown covers area from neck to knees, arms to the end of wrists and wrapped around the back.
  - Tie behind with a simple knot that can be easily untied.
- Perform hand hygiene and don respiratory protection.
  - Use N95 or higher mask/respirator.
  - Secure ties at middle of head and neck.
  - Fit flexible nose piece over the bridge of the nose.
  - Fit snuggly to face and chin.
  - Perform fit test: Mask collapses inward on inspiration and expands without a leak on exhalation.
- Hand hygiene and don goggles.
- Hand hygiene and don head cover, balaclava. Ensure that the sides of the goggles, the ears and nape of neck are covered. Ideally, the neck should be covered as well.
- Hand hygiene and don second pair of gloves. Extend to cover wrist. No skin should be exposed.
- Hand hygiene and don face shield.

Healthcare workers doffing of PPE

- Careful doffing is extremely important to prevent contamination.
- Perform hand hygiene on outer pair of gloves.
- Remove gown (Remember that the front of the gown and the arms are the most contaminated areas). Undo ties and carefully pull the gown down from neck and shoulders. Turn gown inside out.
- Outside gloves are removed with the gown as it reaches the wrist and hands.
- Carefully roll the gown with inside facing outwards into a bundle and discard.
- Perform hand hygiene on inner gloves using alcohol sanitiser.
- Remove face shield (remember that front is contaminated).
- Take off head covering – grasp from behind and carefully lift from the back of the head and into waste packet.
- Perform hand hygiene on inner gloves with alcohol sanitiser.
- Carefully remove goggles.
- Hand hygiene.
- Remove overshoes.
- Hand hygiene.
- Exit doffing area and then remove N95 mask/respirator.
- Hand hygiene.
- Remove inner pair of gloves.
- Maintain these precautions throughout the entire perioperative period.

Table V: Precautions for common pathogens

Pathogen	Level of precautions
HIV	Standard, post-exposure prophylaxis
Hepatitis (viral)	Standard precautions
Human Papilloma Virus (HPV)	Standard, droplet and airborne if plume from cauterisation of airway papillomas or genital warts are present
Mycobacterium tuberculosis	Standard and airborne precautions
Multidrug-resistant organisms Meticillin resistant *Staphylococcus Aureus* (MRSA) Vancomycin-resistant enterococci (VRE) Extended spectrum beta-lactamase-producing organisms (ESBL)	Standard and contact precautions
Neisseria	Standard and droplet precautions
Clostridium difficile	Standard and contract precautions
Haemophilus influenza	Standard and droplet precautions
Influenza	Standard and airborne precautions
Coronaviruses	Standard, droplet and airborne precautions

Chapter 3: General principles

General infection control principles are as follows:

- A named senior member of the anaesthesia staff should be appointed at each hospital to liaise with the infection control team and the occupational health and safety department. This is to ensure establishment of and compliance with best practice standards in infection control in all areas of anaesthetic practice.[9]
- Systems must be established for regular training of healthcare workers in infection control. This must be coupled with monitoring and regular auditing of infection control practice to guard against apathy and poor compliance among staff.[7]
- Teaching and training programmes in the practice of anaesthesia should integrate and promote infection control practices as a fundamental part of the curriculum and the speciality.[5]
- Staff outside the theatre suite dealing with "anaesthetic" equipment, such as laryngoscopes and self-inflating resuscitation devices, need guidance on their decontamination. Particular areas of vulnerability include casualty, the obstetric delivery suite and neonatal intensive care units (NICUs).
- The manufacturer's recommendations should always be consulted to determine the compatibility of the respective piece of equipment with decontamination procedures and disinfectants.
- Changing to single-use anaesthesia devices is the best choice in the prevention of cross-infection. However, care must be taken when choosing a single-use device for an institution. There is a wide range of cheap disposable anaesthesia devices, some of inferior quality. Questions should always be asked as to whether or not the chosen device is fit for the purpose and is an evidence-based choice. Complications, including hypoxia, have been described from the use of inferior, single-use anaesthetic equipment.[11]

Spaulding's classification[12-15]

Spaulding developed a classification of instruments into "critical", "semi-critical" and "non-critical". This classification is more than 50 years old and is used to determine the level of disinfection for certain equipment and items.

Critical instruments

Critical instruments have an increased risk of contamination. It includes those instruments that are in contact with sterile sites within the body or the intravascular system. Critical instruments should be subjected to sterilisation such as pressurised steam or low temperature methods such as ethylene oxide gas. Examples include surgical instruments, implants and sterile catheters. With the advent of multidrug-resistant and aldehyde-resistant organisms there has been a call to also include other semi-critical instruments into the category of critical instruments. One such item is endoscopes. The design is flawed with lots of crevices that may retain microorganisms. A study by Ofstead et al.[16] showed that almost 50% of endoscopes retained moisture that can aid in the development of a biofilm. In addition, 71% of endoscopes showed microbial growth and cultured organisms such as *Stenotrophomonas maltophilia* and *Citrobacter freundii*.

Other items that are relevant to the anaesthetist include airway instruments such as laryngoscope blades and handles as well as forceps. Airway instrumentation is often associated with trauma to the mucosa. Several studies have demonstrated occult blood on these items. These instruments thus meet the criteria for critical instruments.[17]

Semi-critical instruments

These instruments only come into contact with intact mucosa and intact skin. Examples given under this category include endoscopes, respiratory equipment and cystoscopes. It is, however, interesting to note that most of these items are now being considered "critical instruments".

Non-critical instruments

Instruments that only come into contact with intact skin. Divided into patient non-critical items such as blood pressure cuffs and oximetry probes and environmental non-critical items such as cot sides, theatre trolleys and uncontaminated floors.

Chapter 4: Safe injection and drug administration practices

Needles and syringes[9,18-20]

- Needles and syringes are sterile items, intended for single-patient use only.
- A syringe and needle should be considered contaminated after contact with a patient, infusion bag or administration set, and must only be used for that patient.
- Medication should not be administered to different patients from the same syringe, even if a new sterile needle is used for each patient. Changing the needle, but not the syringe, is unacceptable practice.
- A syringe must not be reused, or a used syringe reinserted into a medication vial or solution bag or container, e.g. a saline flush or phenylephrine bag, even if it is for use in the same patient.
- A used needle must not be reinserted into a multiple-dose vial or solution bag or container, e.g. a saline flush or phenylephrine bag, even if it is for use on the same patient.

- The presence of a non-return valve (one-way valve) or the use of a syringe driver or infusion pump does not permit the reuse of syringes or their contents.
- Before use, prepared syringes should be capped to avoid contamination.
- After use or at the end of the anaesthetic, used syringes and needles should be discarded appropriately.
- Syringes must never be stored nor transported in clothing or pockets.
- The presence of a non-return valve (one-way valve) in the infusion set does not prevent the blood contamination of syringes or needles.

Preservative-free (single-dose) ampoules or vials[9,21]

- Preservative-free (single-dose) ampoules or vials are single-dose, single-patient items.
- Do not give drugs from preservative-free vials or ampoules to multiple patients or save the remaining contents for later use.
- Use of single-dose vials is preferred whenever possible over the use of multi-dose vials for parenteral medications.
- They must be disposed of after the drug dose has been drawn up and not reused for other patients.
- Cleanse the vial's rubber septum before entering, or the neck of glass ampoules before breaking, with an alcohol swab. Allow to dry before entering or breaking the vial or ampoule.

Multi-dose vials[9,18,21]

- Use of single-dose vials is preferred whenever possible over that of multi-dose vials for parenteral medications.
- The date and time that a multi-dose vial is opened should be indicated on the vial. If multi-dose vials must be used, then cleanse the vial's rubber septum with an alcohol swab and allow it to dry before entering the vial. A new sterile needle and syringe must be used each time the vial is entered. It must be stored in a clean area between patients, in accordance with the manufacturer's recommendations, in order to prevent cross-contamination from items that have already been used. It must be discarded if there is suspicion that sterility has been compromised. Never leave a needle, cannula or spike device (even if it has a one-way valve) inserted into a medication vial rubber stopper because it leaves the vial vulnerable to contamination.

Infusions, administration sets or items in contact with the vascular system or other sterile body compartments[9,18,20]

- Infusions, administration sets or items that are in contact with the vascular system or other sterile body compartments are for single-patient use. They should be discarded after use.
- Bags or bottles containing intravenous (IV) solution should never be used as a common source of supply for more than one patient, e.g. phenylephrine solutions and saline bags for flushing.
- Never use cannulae or spiking devices, even with a non-return valve, to remove fluid from infusion bottles or bags for multiple uses or patients.
- Use single-dose, single-use containers for flush solutions.
- Aseptic techniques should be used when preparing infusions and breaks or taps in the lines kept to a minimum.
- Always clean IV injection ports with alcohol and allow to dry before use. Cover with a sterile cap after use.
- IV cannula caps or bungs are not to be collected for reuse on other patients. These are single-patient, single-use items.
- Both the syringe and the needle or cannula must be sterile when any medication vial or solution is accessed.
- Propofol should be discarded after six hours of ampoule opening. For continuous IV infusions in the ICU, both the tubing and any unused propofol must be discarded after 12 hours.

Non-injectable items[9]

- Examples include topical drugs, ointments and lubricating gels.
- Ideally, these items should be single-patient, single-use.
- If single-use practice is not possible, utmost care should be taken to avoid self-contamination of these items.
- If contamination is suspected or confirmed, the item should immediately be discarded.

Rationale

The silent and insidious, yet devastating, epidemic of unsafe injection and drug administration practice is plaguing healthcare systems worldwide.[22]

In general, evidence has underestimated reality. A possible explanation is that the pathogens are invisible on the equipment. Also, as HIV, hepatitis B and hepatitis C infections may be clinically silent for a period, an iatrogenic causal association is difficult to establish. Using comparative risk assessment and mathematical modelling, in 2000, the Global Burden of Disease Study estimated that contaminated injections accounted for approximately one in three new hepatitis B virus infections, 40% of new hepatitis C infections and 5% of new HIV infections.[23] Approximately 260 000 HIV/acquired immune deficiency syndrome (AIDS) cases, two million hepatitis C infections and 21 million hepatitis B infections per annum are estimated to occur as a result of the reuse of syringes and needles.[23] In 2010, the Association for Professionals in Infection Control and Epidemiology noted that in the previous decade in the USA alone, unsafe injection, infusion and vial practices resulted in more than 35 outbreaks of viral hepatitis. During this period, more than 100 000 patients were exposed to infectious hepatitis. Anaesthetists were implicated in most of these outbreaks.

Recent observations in South African hospitals and clinics revealed routine failures in injection practice.[24] At least one medical injection in five was noted to be administered with a used syringe or needle. The authors concluded that using financial constraints to justify unsafe injection practices was

"ethically indefensible". Moreover, a recent study published in the *Southern African Journal of Anaesthesiology and Analgesia* showed an unacceptably high prevalence of the reuse of single-patient syringes and spinal fentanyl ampoules by anaesthetists at regional, tertiary and central hospitals in KwaZulu-Natal.[5]

Injection safety is every provider's responsibility. It is especially important to remember that when injecting medication into sterile sites, such as neuraxial techniques, there is no margin for error.

Changing a needle (or cannula) and reusing the syringe is extremely dangerous. Negative pressure is generated when a needle is removed from a syringe, producing a siphoning effect that aspirates the needle contents into the syringe.[19]

IV administration tubing becomes contaminated with blood when backflow occurs during blood sample aspiration, or by accidental gravitation or from a blood transfusion. IV tubing and valves are not sufficient to prevent the backflow and contamination of injection devices. Blood has a higher specific gravity than IV solutions, so passive backflow against forward flowing fluid is possible. Infectious blood-borne organisms may be present, even if blood is not visible in the tubing. Injection at the most distal port from the IV cannula does not prevent contamination of the syringe.[19]

Single-use fentanyl ampoules should not be used on multiple patients undergoing neuraxial anaesthesia. Infectious complications of neuraxial anaesthesia include, but are not limited to, meningitis, encephalitis, and spinal and epidural abscesses.[25] Potential pathogens, such as bacteria from airborne contaminants, nonsterile glass fragments, or failure to use an aseptic technique, may contaminate open, partially used ampoules.[26]

Cases of nosocomial bacterial and viral infections, including fatal hepatitis B infection and fatal bacterial meningitis, have been linked to the use of contaminated multi-dose vials.[27,28] Viable viruses were found to survive for at least 24 hours in one study, while bacteria and endotoxins were also found in contaminated multi-dose vials.[28]

Injection ports are a route of entry for microorganisms into the vascular system or other sterile body compartments.[29]

Sharing saline bags as flushing or medication solutions, e.g. phenylephrine, is not recommended. Case reports include the iatrogenic spread of hepatitis C to at least 99 patients at an outpatient clinic as a result of disposable syringe reuse and the contamination of shared saline bags.[30]

Bacterial and fungal infections in postsurgical patients have been linked to contaminated infusions used on multiple patients. Propofol carries a high risk of contamination and cases of postsurgical sepsis and deaths from contaminated infusions have been reported.[25] Propofol is manufactured in a nutrient-rich emulsion, containing glycerine, soybean oil and egg phospholipids, and bacterial growth has been documented at six hours, despite the use of preservative.[31] Therefore, manufacturers have recommended that propofol be discarded six hours after ampoule opening, and for continuous IV infusions, the tubing, container and any unused propofol after 12 hours.[32,33]

Chapter 5: Hand hygiene

Hand washing is one of the most effective infection control practices.

Indications for hand hygiene:

- Before and after direct patient contact.
- Before putting on sterile gloves.
- Contact with body fluids, mucous membranes, open skin and wound dressings.
- Before making contact with a clean site after touching a contaminated site.
- After touching a high-touch environmental surface/equipment near the patient.
- After removing gloves.
- Before eating or drinking.
- After using the bathroom.

Gloves do not fully protect against hand contamination.

Plain soap (non-antimicrobial): used for routine hand washing, when hands look dirty. It will only remove loosely adherent transient bacteria through mechanical friction. It will not decontaminate the hands.

Antimicrobial soap: used if there has been visible hand contamination with blood or body fluids. Chlorhexidine has adequate cover against Gram-positive, Gram-negative organisms and viruses but poor cover against mycobacterium and fungi. Iodine compounds have a slightly decreased action against fungi and good cover against all the other microorganism groups. Both chlorhexidine and iodine compounds have an intermediate onset of action and thus need adequate contact time. Chlorhexidine retains its efficacy in the presence of blood and has a lower incidence of adverse skin reactions.

Alcohol-based hand rubs can be used provided the hands are not obviously dirty or contaminated with proteinaceous material. If an alcohol hand rub is used, it is important to keep the hands and forearms wet during the whole procedure. Using inadequate volumes of alcohol-based rub (0.2–0.5 ml) is as efficient as washing with plain soap and water. Approximately 15 ml of alcohol-based rub is required. Roughly one minute should be spent rubbing the forearm. Thereafter, hands and fingers should

be rubbed in the same manner as for hand washing. Alcohol has good cover against Gram-positive and Gram-negative organisms and also against mycobacteria and viruses. It also has a fast onset of action and should be allowed to evaporate. It has virtually no activity against spores and protozoal oozytes. Physical hand washing with antimicrobial soap and vigorous washing and rinsing is important for spore-forming organisms, such as *Clostridium difficile* and *Bacillus anthracis*.

Nails: Artificial nails and nail polish should not be used in the operating rooms or ICU. Fingernails should be kept short and clean. Avoid using nail brushes during hand washing as they damage the skin and therefore increase the risk of contamination by microorganisms.

Non-sterile gloves should be worn whenever contact with blood, body fluid, mucous membranes, non-intact skin and potentially infectious materials is anticipated. They need to be removed as soon as possible, and therefore be changed between different procedures on the same patient. They should not be reused. Gloves must be removed before touching equipment if they were in contact with the patient. Curtains, clinical notes, pens, computer keyboards, and cellular and landline telephones must not be touched with contaminated gloves.

Alcohol-based hand rubs used on gloved hands can be considered in settings with inadequate supply of gloves to meet the above requirements. This practice may potentially degrade the quality of the gloves, making them prone to leakage.

Bare below the elbow: Different societies have conflicting recommendations with regards to coverage of the arms. This contentious issue has led to multiple guideline changes. Unfortunately, studies have had conflicting results. The proponents of "bare below the elbow" argue that garments have a high risk of being contaminated and by removing them as far away as possible from the hands will decrease the risk of contamination of the hands and increase the efficacy of hand washing. Opponents to "bare below the elbow" argue that the skin sheds millions of squamous cells every day that contain both normal skin flora and pathogenic microorganisms. By covering up the arms to the wrists, the load of squamous cells entering the environment will be decreased. False nails, nail polish, wristwatches and stoned rings should not be worn.

Simple hand washing has been shown to be one of the most effective infection control practices in everyday practice in protecting anaesthesiologists and patients from colonisation and/or infection with microorganisms.[34-36]

Rationale

Our hands carry a high count ($3.9 \times 104–4.6 \times 106$ of colony-forming units [CFUs]/cm^2) of resident and transient bacteria.[37-40] The function of the resident flora is microbial antagonism by competing for nutrients. Resident flora are usually not associated with healthcare-associated infections, but they may cause infection when introduced into sterile body cavities.[37,41]

Transient flora are the more problematic pathogens. Patients harbour pathological bacteria in septic wounds, and also on intact skin. Since on average humans shed approximately 106 skin squames per day,[42] these pathological microbes also contaminate the environment around the patient. Healthcare workers can contaminate their hands by simple "clean" procedures, such as feeling a patient's pulse, or by touching the bed or the patient's file.[37,43-51] In one study,[52] 52% of healthcare workers who entered vancomycin-resistant enterococci (VRE) infected patients' cubicles and whose hands were free from VRE, were contaminated by the time they exited the cubicles. These healthcare workers had not touched the patients at all and were contaminated purely by interacting with the surrounding environment. There was a 70% contamination rate for the healthcare workers who touched patients.

Gloves do not fully protect against contamination.[53] When high-risk procedures such as nappy changes were examined, there was only a 50% reduction in contamination when gloves were used. In a study by Hayden et al.[52] on VRE contamination, 3% of VRE-negative healthcare workers who were wearing gloves were contaminated. This was 86% lower than their ungloved counterparts. Ehrenkranz and Alfonso[54] cultured the moisture found on the inside of used gloves after nursing staff had touched the groins of patients infected with *Proteus mirabilis,* and found up to 600 CFU/ml of the organism. Although gloves are a useful adjunct to hand hygiene, they are not a substitute for proper hand decontamination.

Various studies have shown that microorganisms can survive for prolonged periods. Noskin et al.[55] showed that VRE can survive for more than 60 minutes on gloved and ungloved hands, and in the environment. This was also true for *Enterococcus feacalis*, *E. faecium*, *Pseudomonas aeruginosa* and *Burkholderia cepacia* when used in a study to show contamination by handshaking. When the bacteria were suspended in saline, contamination occurred up to 30 minutes later. However, if these bacteria were suspended in sputum, contamination occurred up to 180 minutes later.[56]

Despite this compelling evidence that hands are an important vector for cross-contamination, in general, compliance rates in healthcare workers are low. It is difficult to determine an exact figure because when overt observers are used in studies, the compliance tends to be higher because of the so-called "Hawthorne effect".[57] If you know that you are being watched, you will tend to be more compliant. Covert observers may not witness every opportunity for hand washing, and they may also be noticed by healthcare workers, which again will lead to the Hawthorne effect. In one study, the Hawthorne effect led to a 55% increase in compliance.[58] Overall, between various studies, the compliance rate remained below 50%.[59-61]

A study by Biddle and Shah[62] showed that indications for hand hygiene according to the WHO 5 Moments of Hand Hygiene reached 54 episodes per hour in high task density periods such as challenging airway situations, complicated emergence or the haemodynamically unstable patient. In 82% of incidents where

hand hygiene should have occurred, there was failure to do so by all providers.

Munoz-Price et al.[63] showed that placing an alcohol-based hand rub on the anaesthetic work station improved compliance to hand hygiene.

The quality of hand decontamination also remains a problem. The amount of time spent on hand washing, the amount of decontaminant used and the type of decontaminant all play an important role. Noskin et al.[55] found that after an inoculum of VRE, hand washing for five seconds with water alone made no difference to the degree of contamination. When two soaps were used for five seconds, there was still 1% recovery of the initial inoculum. A time of 30 seconds was needed for complete decontamination. In laboratory studies by Larson et al., it was found that one ml of alcohol-based hand rub or liquid soap had lower bacterial reduction rates than three ml of the product.

Indications for hand hygiene are as follows:[34,36]

- Before and after direct patient contact.
- Before putting on sterile gloves.
- Contact with body fluids, mucous membranes, open skin and wound dressings.
- Before making contact with a clean site after touching a contaminated site.
- After touching a high-touch environmental surface near the patient.
- After removing gloves.
- Before and after eating or drinking.
- After using the bathroom.

Recommendations

Plain soap (non-antimicrobial)

Plain soap (non-antimicrobial) should be used for routine hand washing when hands look dirty. It will only remove loosely adherent transient bacteria and will not decontaminate the hands.[54,64,65]

Soap (antimicrobial)

Antimicrobial soap should be used if there has been visible hand contamination with blood or body fluids.

Alcohol-based hand rubs

Alcohol-based hand rubs[37] can be used provided the hands are not obviously dirty or contaminated with proteinaceous material. If an alcohol hand rub is used, it is important to keep the hands and forearms wet during the whole procedure.[66] Using inadequate volumes of alcohol-based rub (0.2–0.5 ml) is as efficient as washing with plain soap and water.[67,68] Approximately 15 ml of alcohol-based rub is required. Roughly one minute should be spent rubbing the forearm. Thereafter, hands and fingers should be rubbed in the same manner as for hand washing. The hands should be kept above the level of the elbows. When choosing an alcohol-based hand gel, it is very important to ensure that it complies with the test standard EN 12791[58] for hand rub formulations. Many commercially available hygienic hand rubs do not meet the standard EN 1500[69] which is similar to the EN 12791. Alcohol-based hand rubs should be easily accessible to improve compliance. They should be placed close to the anaesthesia workstation or close to the doors to the operating theatre. Wall-mounted dispensers will also increase compliance.[70,71] Dispensers should be at least 2.5 cm away from ignition sources as there is a small risk of fire.

Alcohol has virtually no activity against bacterial spores and protozoal oozytes.[37] If there is a high risk of these pathogens, then alcohol should not be used as the sole agent for decontamination. Physical hand washing with water-based washing and rinsing is important for spore-forming organisms, such as *Clostridium difficile* and *Bacillus anthracis*.

Nails[37,39,72-75]

Despite low-quality evidence and some studies showing that nail polish or gel nails do not increase the bacterial burden, it is recommended that artificial nails and nail polish should not be used in the operating rooms or ICU. Fingernails should be kept short and clean. Avoid using nail brushes during hand washing as they damage the skin.

Sterile gloves

Sterile gloves must be worn if an invasive procedure is performed or if there is contact with sterile sites. Gloves should be of high quality to ensure patient and healthcare worker safety.

Non-sterile gloves

Gloves should be of high quality to ensure patient and healthcare worker safety. Non-sterile gloves should be worn whenever contact with blood, body fluid, mucous membranes, non-intact skin and potentially infectious materials is anticipated. They must be put on immediately before patient contact. They need to be removed as soon as possible, and thus be changed between different procedures on the same patient. They should not be reused. Gloves must be removed before touching equipment if they were in contact with the patient. Curtains, clinical notes, pens, computer keyboards, cellular and landline telephones must not be touched with contaminated gloves. Hand washing should be performed as soon as possible. Double gloves can also be worn, especially during airway manipulation where the anaesthetist's hands may become contaminated with respiratory secretions. The outer glove may be removed before touching environmental surfaces such as setting the ventilator. Gloves should not be a substitute for hand washing as there can be a degree of glove leakage during use and self-contamination when removing the gloves. Powdered and polythene gloves should not be used.

Some institutions, however, may have limited supplies of gloves that will make it challenging to change gloves as per the above indications. Current data on whether gloved hands can be disinfected with alcohol-based hand rubs are inconclusive. The exact effect of such a practice on the integrity of gloves is unknown. A study by Gao et al.[76] investigated the influence of

alcohol-based hand rubs on the tensile strength of gloves. If tensile strength can indeed be extrapolated to glove integrity, then alcohol does not seem to affect the gloves. A limitation of the study is that only thirteen brands of latex and nitrile gloves were studied. Resource-constrained facilities may opt for cheaper gloves that may not withstand the effects of alcohol-based hand rubs. Using alcohol-based hand rubs on gloves is deemed to be better than no hand hygiene at all and should be considered when gloves are in short supply.

Bare below the elbow/Arms fully covered[77,78]

Different societies have conflicting recommendations with regards to coverage of the arms. This contentious issue has led to multiple guideline changes. Unfortunately, studies have had conflicting results. The proponents of "bare below the elbow" argue that garments have a high risk of being contaminated and removing them as far away as possible from the hands will decrease the risk of contamination of the hands and increase the efficacy of hand washing. Opponents to "bare below the elbow" argue that the skin sheds millions of squamous cells every day that contain both normal skin flora and pathogenic microorganisms. By covering up the arms to the wrists, the load of squamous cells entering the environment will be decreased. Studies looking at "bare below the elbows" failed to prove that it actually made a difference in colony-forming units when fingertips were cultured.[77,78] A study by Markel et al.[79] showed a decrease in microbial contamination and particle counts when a long-sleeved garment and sterile gloves were used in mock skin preparations. The biggest decrease was for particles more than 5 µm in size.

False nails, nail polish, wristwatches and stoned rings should not be worn.

Cuts and abrasions on healthcare workers

Cuts and abrasions on healthcare workers must be covered with waterproof dressings.

High-touch environmental surfaces

The most common high-touch environment surfaces are the adjustable pressure-limiting valve and the agent concentration dial.

Hand washing technique (three-stage technique)[37,39]

The three-stage hand washing technique is as follows:

- *Stage 1 – Preparation:* Wet hands thoroughly under tepid running water before applying liquid soap or antimicrobial solution. Ensure that the hand wash solution comes into contact with all surfaces of the hand, including the wrists. Avoid very hot water as it causes skin damage.
- *Stage 2 – Washing and rinsing:* Rub hands together vigorously for a minimum of 15 seconds. Pay particular attention to fingertips, thumbs and areas between the fingers. Hands should then be thoroughly rinsed.
- *Stage 3 – Drying:* Dry thoroughly with good-quality disposable paper towel. When drying the hands, they should be patted, rather than rubbed, as rubbing leads to small cracks in the skin.

Chapter 6: Anaesthetic equipment decontamination

Management of reusable equipment

- Reprocessing refers to infection control procedures for removing and inactivating microorganisms on reusable patient-care equipment.
- Reprocessing of reusable patient-care equipment includes cleaning, disinfection, and sterilisation.
- The choice of reprocessing method must be guided by the instrument manufacturer's recommendations.
- Factors that need to be considered are compatibility among equipment components and materials, chemicals to be used, heat and pressure tolerance of the equipment, and time and temperature requirements of the reprocessing methods.

Definitions and classifications

Spaulding classification[12-15]

Spaulding developed a classification of instruments into "critical", "semi-critical" and "non-critical". This classification is more than 50 years old and is used to determine the level of disinfection for equipment items.

Critical instruments

- Critical instruments have a high risk of contamination.
- This includes those instruments that are in contact with sterile sites within the body or the intravascular system.
- Critical instruments should be subjected to sterilisation such as pressurised steam or low temperature methods such as ethylene oxide gas.
- Examples include surgical instruments, implants and sterile catheters.
- With the advent of multidrug-resistant and aldehyde-resistant organisms there was a call to also include other semi-critical instruments into the category of critical instruments. One such an item is endoscopes. The design is flawed with lots of crevices that may retain microorganisms. A study by Ofstead et al.[16] showed that almost 50% of endoscopes retained moisture that can aid in the development of a biofilm. In addition, 71% of endoscopes showed microbial growth and cultured organisms such as *Stenotrophomonas maltophilia* and *Citrobacter freundii*.
- Other items that are relevant to the anaesthetist include airway instruments such as laryngoscope blades and handles as well

as forceps. Airway instrumentation is often associated with trauma to the mucosa. Several studies have demonstrated occult blood on these items. These instruments thus meet the criteria for critical instruments.[17] These items require cleaning and disinfection followed by sterilisation as required and as per manufacturer's recommendations.

Semi-critical instruments

- Semi-critical instruments have an intermediate risk of contamination.
- These instruments only come into contact with intact mucosa and intact skin.
- Examples given under this category include endoscopes, respiratory equipment and cystoscopes. It is however interesting to note that most of these items are now being considered "critical instruments".

Non-critical instruments

- Non-critical equipment has a low risk of contamination.
- These instruments only come into contact with intact skin.
- Divided into patient non-critical items such as blood pressure cuffs and oximetry probes and environmental non-critical items such as cot sides, theatre trolleys and uncontaminated floors.
- Cleaning and drying are usually adequate.

Laryngoscopes

Laryngoscope blades[14,80]

Contaminated anaesthetic equipment has been implicated in the nosocomial transmission of infectious diseases.[81] Options for the reprocessing of laryngoscope blades include:

- *Use of disposable (single-use) laryngoscope blades (DLBs) (preferred):* The metal type only must be used. They should be discarded after single use. DLBs should not be reused, even after sterilisation.
- *The sterilisation of reusable laryngoscope blades (RLBs):* The light intensity of all RLBs that are steam sterilised should be monitored. Handling and storage are important.
- *High-level disinfection (HLD):* There are significant concerns about the use of HLD in a South African setting when decontaminating RLBs. Evidence of poor compliance with HLD protocol has been documented, and there is a significant margin for human error in the HLD protocol. If it is used, hospitals must have a specific step-by-step instructional protocol in print that is well understood. Decontamination should occur in a specific designated area, away from patients and other healthcare workers. Frequent in-service training of anaesthesia nurses on HLD must be conducted. The decontamination of RLBs should be monitored and audited for compliance.
- No other method of disinfection, e.g. chlorhexidine and alcohol, should be used to decontaminate RLBs.

Rationale

Laryngoscope blades have been implicated in the transmission of nosocomial disease.[82-84] Other studies have identified RLBs as potential vectors for the transmission of methicillin-resistant *Staphylococcus aureus*, *P. aeruginosa, Serratia marcescens* and other pathogenic microorganisms.[4,85] Further studies have identified the presence of blood (occult and/or visible blood) on RLBs deemed to be ready for use.[6,17,86,87] These include a study conducted in the regional, tertiary and central hospitals in a province of South Africa which found blood (visible or occult) and/or visible organic material contamination of RLBs of 80%.[6] Staff from 11 of the 15 studied hospitals claimed that the blades were decontaminated by HLD.[7]

If inanimate objects become contaminated with hepatitis B virus and are not appropriately decontaminated, these objects may contribute to disease transmission for periods of up to one week and possibly longer.[88] Hepatitis C has been shown to retain infectivity for several days at room temperature. Wenzel and Edmond[89] acknowledged that instruments are sources of pulmonary infections with Gram-negative organisms, such as *P. aeruginosa* or *S. marcescens*, reflecting an inanimate environmental reservoir. They concluded that if 1–5% of bronchoscopic procedures were performed on patients with tuberculosis, and if each was followed by a second procedure with the same scope, 460–2 300 patients might become exposed to the pathogen each year if only 10% of the scopes were contaminated. One of the most compelling reasons for re-evaluating the cleaning, disinfection and sterilisation techniques of airway management equipment derives from the report by Agerton et al.[90] on outbreaks of tuberculosis following bronchoscopic procedures.

Blood contamination of reusable laryngoscope blades

Traditionally, the RLB was classified by the American Society of Anesthesiologists (ASA) as a "semi-critical" medical device using the Spaulding classification scheme.[34,91] According to this scheme, medical devices that directly or indirectly contact mucous membranes or non-intact skin, without ordinarily entering sterile tissue or the vasculature, are classified as semi-critical devices, for which this scheme prescribes at least HLD or sterilisation.

Concerns about this classification of RLBs were raised both locally and abroad.[6,9] Bleeding in the mouth following routine laryngoscopy has been well described, as well as the contamination of laryngoscope blades with this blood.[92] This implies penetration, and not merely contact with the mucosal tissue by the blade. The laryngoscope blade, when used during dental, maxillofacial and otorhinolaryngology surgery, may also become grossly contaminated with blood. The presence of blood contamination from prior patients may facilitate the nosocomial transmission of the hepatitis B and C viruses, HIV and other blood-borne pathogens, the risk of which is difficult to ascertain owing to the paucity of documented cases and ethical constraints in performing such studies.

However, mucosal membrane contact with blood tissue, or other body fluids that are potentially infectious, as well as percutaneous injury, are defined as exposure which places patients at risk of acquiring HIV infection.[93] In retrospective case-control and post-exposure prophylaxis studies on HIV, an increased risk of HIV infection was associated with exposure to a large quantity of blood from the source, a device visibly contaminated with the patient's blood, and a deep injury. Other factors included a source patient with AIDS, patients with a high viral load and injury with a hollow-bore needle.[93] Furthermore, hepatitis B is approximately 100 times more transmissible than HIV. Contaminated RLBs that may or may not cause a traumatic mucosal breach pose a risk of transmission of hepatitis B, hepatitis C and HIV. Therefore, RLBs should rather be considered to be critical items, rather than semi-critical ones, especially in countries with high endemic levels of HIV and hepatitis B positivity. Several countries in Europe, including Britain and the Netherlands, have changed the decontamination of RLBs from HLD to sterilisation after each use or adopted the use of suitable disposables.[9]

The rigidity of the high-level disinfection protocol

There are three essential stages in HLD:[94,95]

- *Cleaning:* Removal of visible contamination from surfaces with water and friction, e.g. the use of a brush and fluidics, i.e. fluid under pressure, together with enzymatic products. Sequestered organic material poses the greatest risk of cross-contamination for patients as it impedes the effectiveness of these cleaning processes by reacting chemically with the germicide, and/or by forming a protective physical barrier for microorganisms.
- *Immersion in a high-level disinfectant:* Immersion in a high-level disinfectant must take place, i.e. ortho-phthalaldehyde (Cidex® OPA) and glutaraldehyde (Cidex®). The duration of immersion should be in accordance with the manufacturer's recommendations, with exposure times varying between 8–45 minutes at 20–25 ºC.
- *Removal of the disinfectant:* Removal of the disinfectant is achieved by adequate rinsing with water.

All three steps are fundamental to the effectiveness and safety of HLD.

The effectiveness and safety of HLD is compromised by:

- Human factors, i.e. owing to complacency and the ignorance of staff, staff shortages and staff turnover.
- Insufficient time allocated to the decontamination process.
- Inadequate numbers of available laryngoscope blades per theatre to allow for compliance with the minimum duration of exposure to the disinfectant in order to achieve effective HLD.

Several of the factors were identified or hypothesised by a study conducted in South Africa, where 60% of the hospitals that claimed to practice HLD were actually noncompliant with the HLD protocol, owing to noncompliance with one or more of the three steps of the process, for example, the omission of cleaning prior to immersion.[7]

Variations and inconsistencies in reprocessing guidelines and practices can result in ineffective reprocessing, confusion among healthcare staff members, inconsistent and inadequate standards of care, and an increased risk of patient-to-patient disease transmission.[96] Moreover, there can be no margin for human error in South African hospitals in this regard because of the risk of transmitting HIV and hepatitis.

The occupational hazard of high-level disinfectants[14,95]

Commonly used high-level disinfectants in South Africa include ortho-phthalaldehyde (Cidex® OPA) and glutaraldehyde (Cidex®). Ortho-phthalaldehyde (Cidex® OPA) is preferred to glutaraldehyde (Cidex®) for use as a disinfectant. Cidex® OPA has excellent stability over a wide pH range (pH 3–9), is not a known irritant to the eyes and nasal passages, does not require exposure monitoring, has a barely perceptible odour and requires no activation. A potential disadvantage of Cidex® OPA is that it stains proteins grey, including unprotected skin, and thus must be handled with caution with the use of gloves and protective clothing. Cidex® OPA residue remaining on inadequately water-rinsed equipment can stain the patient's mouth. Allergic reactions have been reported in urology patients undergoing repeated cystoscopies with scopes reprocessed with Cidex® OPA. In April 2004, the manufacturer of OPA alerted the medical community about patients who reportedly experienced an anaphylaxis-like reaction after cystoscopy with scopes repeatedly disinfected with Cidex® OPA.

Glutaraldehyde (Cidex®) causes serious occupational hazard concerns. Cidex® OPA solution should be used in a well-ventilated area and stored in closed containers with tight-fitting lids. Healthcare personnel can be exposed to elevated levels of glutaraldehyde (Cidex®) vapour when equipment is processed in poorly ventilated rooms, when spills occur, when glutaraldehyde solutions are activated or changed, or when open immersion baths are used. Acute or chronic exposure can result in skin irritation or dermatitis, mucous membrane irritation (i.e. the eyes, nose and mouth), or pulmonary symptoms. Epistaxis, allergic contact dermatitis, asthma and rhinitis has also been reported in healthcare workers who had been exposed to glutaraldehyde. Glutaraldehyde exposure needs to be monitored to ensure a safe working environment.

The case for disposable (single-use) laryngoscope blades

Single-use airway equipment is designed to be used once and then discarded. Studies have shown that current techniques for cleaning and decontaminating RLBs are ineffective in removing all remnants of blood. If anaesthetists are not confident that RLBs are appropriately decontaminated, they should use disposable equipment.[97]

Different DLBs are manufactured with different designs and materials. There may be concern about the quality of some of these devices because they are manufactured at a lower cost to justify their disposal.[11] However, metal DLBs offer decreased infection risk with optimal user satisfaction and safety. A multicentre randomised study on more than 1 000 patients for emergency intubation under rapid sequence intubation was

published in 2010. It found that single-use metal blades were more efficient than reusable metal blades. Significantly fewer failed first attempts, and there were fewer poor-grade laryngeal views.[98]

A plastic DLB is reported to be less efficient than a metal reusable blade during rapid sequence induction. This is owing to the increased flexibility of the blade. Jabre et al.[99] recommended that conventional RLBs should be available for difficult intubations, and plastic DLBs used for uncomplicated intubations. Therefore, metal DLBs are recommended.

Interestingly, studies have shown that most clinicians would prefer single-use devices to be used on themselves and their families if they were patients.[11] The use of DLBs removes concerns about decontamination, and particularly the human factors.

The case for sterilisation of reusable laryngoscope blades[95,96]

Sterilisation of RLBs has significant advantages over HLD:

- *Reliability:* Sterilisation has a larger margin of safety, coupled with reliability, consistency and lethality. Sterilisation, in contrast to HLD, significantly removes the "human" element from the process of decontamination.
- *Cost-effective:* Sterilisation is relatively inexpensive. It decreases the need for high-level disinfectants, with the associated need for storage containers and protective gear.
- *Improved efficiency:* It links to improved theatre turnover rates, with less time required for decontamination between cases.
- *Patient benefits:* There is a decreased risk of nosocomial infection and decreased exposure to high-level disinfectants and their residue with sterilisation.
- *Healthcare worker benefits:* Sterilisation reduces the anaesthesia staff workload. It removes occupational health and safety hazards associated with high-level disinfectants.

The challenge with sterilisation is the progressive decrease in the light intensity of the laryngoscope blades that undergo sterilisation. Therefore, the lifespan of the laryngoscope blade is shorter. Reusable fibreoptic laryngoscope blades have been shown to deteriorate with repeated steam sterilisation, eventually becoming less bright.[100-102]

Different brands of laryngoscope blades tolerate automated machine cleaning and steam sterilisation differently, and this needs to be kept in mind when making future purchases.

Handling of the contaminated laryngoscope blade

Gadalla and Fong devised a way of improving the handling of the contaminated laryngoscope blade.[103] First, the anaesthetist puts on two pairs of clean gloves. Induction is carried out, and then as soon as endotracheal tube placement is complete, the blade of the laryngoscope is held in the gloved hand, and one outer glove is peeled off the hand and inverted over the dirty laryngoscope blade. The other outer glove is also removed. The anaesthetist is then left with a clean pair of gloves. This technique ensures that the used blade never comes into contact with other equipment.

The Association of Anaesthetists of Great Britain and Ireland (AAGBI)[9] recommends that the anaesthetist wear gloves during intubation and then place the used blade in a designated receptacle to prevent contamination of surfaces, pillows and drapes.

Laryngoscope handles[95,104-106]

The laryngoscope handle should be decontaminated after each patient. To minimise laryngoscope handle contamination:

- Remove the blade from the handle immediately after use and place the contaminated blade in a receptacle.
- Do not close the contaminated blade on the handle after intubation.
- Consider covering the handle with a new disposable plastic bag for each patient, as described in the rationale. (This does not change the need for HLD or sterilisation). Consider using the specifically devised double-glove, clean induction technique by Gadalla and Fong.[103]
- Decontaminate by sterilisation, or HLD or ILD.

Sterilisation

Sterilisation steps are as follows:

- Send the laryngoscope handle to the central sterile supplies department for sterilisation.
- Batteries must be removed in the OT.
- Adequate numbers of handles per OT should be acquired to allow for this.
- The manufacturer should be consulted to determine compatibility with the type of sterilisation. Steam sterilisation is recommended. If the handle is not sterilisation friendly, it should be replaced.

High-level disinfection

HLD steps are as follows:

- HLD should take place after each patient.
- Batteries should be removed in the OT prior to HLD.

There should be a specific step-by-step instructional protocol in print, that can be easily understood.

- HLD must be monitored and audited for compliance.
- Adequate numbers of laryngoscope handles per OT must be acquired to allow for this.

Intermediate-level disinfection

ILD steps are as follows:

- ILD should take place after each patient.
- Chlorhexidine 2% with alcohol 70% should be used.
- HLD or sterilisation must be employed if there is visible blood or organic material contamination.
- Several articles prefer HLD over ILD, so ILD is not the preferred choice.

- After sterilisation or disinfection, the laryngoscope handle and blade should be checked for function and then package into a sealed plastic bag to prevent recontamination.

Rationale

A recent review of laryngoscope blades and handles as a source of infection concluded that the laryngoscope handle should be considered to be a semi-critical item, at least requiring HLD.[107,108] It does not come into direct contact with the patient's oral mucosa. However, it may become contaminated by the tip of the blade when the blade is folded in the "off" position.[86,109,110] This "contact point" presents a potential route for the patient-to-patient transmission of blood and organisms from the oropharynx. The laryngoscope handle can also be contaminated by the clinician's gloves, by direct contact with surfaces or other anaesthetic equipment, as well as by indirect contact from splashes or airborne pathogens. Microorganisms can then be transmitted to subsequent patients when the clean blade touches the laryngoscope handle, or when the anaesthesia provider's gloves touch a contaminated laryngoscope handle. Studies have found high contamination rates of laryngoscope handles, including drug-resistant organisms.[87,105,110,111]

Traditionally, laryngoscope handles have a knurled surface for good grip. However, the fissures in this surface may provide a protected niche for pathogens[109,112] A greater range and heavier growth of bacterial species was found on the knurled, compared with smooth, surfaces of the handle, suggesting that knurled surfaces may harbour more bacteria than smooth surfaces.[105] Two recent articles called for the revision of inconsistent guidelines for disinfection and the storage of laryngoscope handles prior to use and recommended at least HLD for laryngoscope handles.[96,105]

Williams et al.[105] isolated one or more species of bacteria from 86% of laryngoscope handles examined, despite the use of low-level disinfection. Isolates included organisms such as *Enterococci,* methicillin-susceptible *S. aureus, Klebsiella* spp. and *Acinetobacter* spp. The Medicines and Healthcare Products Regulatory Agency of the UK reported on the death of a patient who developed sepsis from a contaminated laryngoscope handle.[113]

Choice of strategies to prevent cross-infection will be influenced by a cost-benefit analysis, but includes the use of newly sterilised laryngoscope handles for each case, disposable laryngoscope handles or HLD of handles between each case. ILD with chlorhexidine 2% with alcohol 70% has been used. However, the trend in the literature is moving away from ILD to at least HLD. If ILD or HLD are used, sterilising handles on a scheduled basis is recommended, especially if *C. difficile* is encountered.[107]

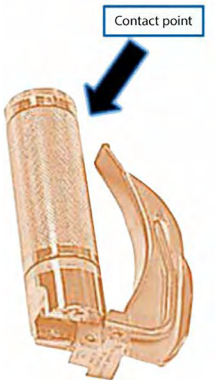

Figure 1: Contact point between the laryngoscope blade and the handle*
* Reproduced with permission[85]

A plastic bag or sheath has been used to cover the laryngoscope handle in some cases, but this practice has been criticised for creating a false sense of security. Full sterilisation procedures or HLD of both the laryngoscope handle and the blade have been advocated.[9,96]

Only one hospital of the 15 studied decontaminated laryngoscope handles after each use in a study that was conducted in regional, tertiary and central hospitals in a province of South Africa.[6] It is imperative that laryngoscope handles are decontaminated between use.[9,114]

Table I: Step-by-step decontamination of laryngoscope blades and handle

Step 1: Transportation from point of use Transport the laryngoscope blade and handle promptly in a suitable container to the designated decontamination area. Dried debris is both difficult to clean and can impede the effectiveness of disinfection or sterilisation. Therefore avoid delays in reprocessing after use.
Step 2: Disassembly in the decontamination area Detach the laryngoscope blade from the handle. Remove the fibreoptic light bundle or the light bulb from the blade if necessary, and/or the batteries or lamp cartridge unit from the handle.
Step 3: Cleaning Clean the blade and handle using enzymatic detergent, a brush and fresh, clean water. Soak the entire blade and handle in the detergent solution, unless contraindicated by the manufacturer's reprocessing instructions. Rinse the blade and handle with a large volume of fresh, clean water. Drying is important as it retards biofilm development.
Step 4: Sterilisation or high-level disinfection Steam sterilise the blade and handle, unless contraindicated in the manufacturer's reprocessing instructions. If steam sterilisation of blade and/or handle is contraindicated, a low-temperature sterilisation process can be considered. Pasteurisation is another recommended method for decontaminating laryngoscope blades and/or handles. If sterilisation of the blade and/or handle is not possible, HLD should be used according to the laryngoscope manufacturer's instructions. Immersion in a high-level disinfectant: ortho-phthalaldehyde (Cidex® OPA) or glutaraldehyde (Cidex®). The duration of immersion should be in accordance with the manufacturer's recommendations. After HLD, rinse the blade and/or handle with a large volume of fresh, clean water. Inadequate rinsing may result in instrument damage or injury to the patient's respiratory mucosa. Do not reuse the rinse water. The blade and/or handle should then be dried with a clean, dry, soft, lint-free cloth. The cloth, dampened with 70% alcohol, may be used to facilitate drying.
Step 5: Transportation, storage, handling and care If the laryngoscope is to be stored prior to use, transport it to the storage area, using care to prevent recontamination. The storage area should be clean and dry. To avoid bacterial colonisation, the blade and handle should not be stored in a closed carrying case, container or kit.

Bronchoscopes

The step-by-step decontamination of bronchoscopes is necessary to prevent both cross-infection between patients and also damage to the scope from debris.

Table II: Step-by-step decontamination of bronchoscopes

Step 1: Pre-cleaning at the bedside Immediately after use wipe the entire endoscope, including any axillary channels and the insertion tube, with gauze soaked with a detergent solution while at the bedside. Place the insertion tube into the detergent solution and suction the detergent up through the instrument channel for several seconds. Finally, clear the channel by suctioning air. If the patient secretions and enzymatic detergents in endoscopic lumens are allowed to dry, they become difficult to remove. Prompt flushing and wiping prevent this.
Step 2: Transporting to the designated decontamination area Used devices must be handled using routine infection prevention and control practices.
Step 3: Leak testing Unless otherwise specified by the endoscope manufacturer, leak testing must be performed after each use, prior to cleaning, to verify the integrity of the endoscope. The leak test should be performed according to the endoscope manufacturer's instructions. The purpose of the leak test is to detect damage to the endoscope. The purpose of the leak test is to minimise damage to parts of the device due to fluid exposure during disinfection. A damaged device must be immediately removed from service and labelled to ensure that it is not used until the device is repaired.
Step 4: Cleaning and rinsing Completely immerse and manually clean before disinfection. Manual cleaning should be performed according to specific manufacturer's instructions for each model. Residual organic material can impede and prevent effective disinfection or sterilisation. Unless otherwise recommended by the manufacturer, the bronchoscope should be completely immersed in a freshly made solution of water and enzymatic detergent that is compatible with the device. The size and diameter of the container/basin should be large enough to prevent undue stress to the bronchoscope. Ensure that all necessary PPE is available for staff. When an enzymatic detergent is used, ensure complete rinsing because the residue may deactivate high-level disinfectants. Brushing and wiping is used to wash debris from the exterior of the scope while it is completely immersed in the detergent solution. The channels and lumens of the endoscope must be flushed and brushed in accordance with the manufacturer's instructions. The brushes used to clean the lumens should be of an appropriate size and must be inspected before use. Common reprocessing errors are the use of an incorrect brush for a particular channel, or the use of a damaged or contaminated brush. The endoscope, and all removed accessories, must then be thoroughly rinsed with clean, fresh water to remove residual debris and detergent. The rinsing water should be approximately three times the volume of the lumen. Remove excess rinsing water to prevent dilution of the subsequently used liquid chemical disinfectant.
Step 5: High-level disinfection or sterilisation Bronchoscopes should be sterilised preferably, if the manufacturer's instructions allow it. (Sterilisable bronchoscopes are available). If sterilisation is not permitted in the instructions, the bronchoscope should receive HLD. If the bronchoscope is not being used on a routine basis, it should be reprocessed prior to use. Manual HLD should be carried out as follows: Completely immerse the previously cleaned and rinsed endoscope and removable parts in a basin of high-level disinfectant. Ensure that the basin is large enough to accommodate the scope without undue coiling and stress, and that it has a tight-fitting lid to contain chemical vapours. Inject the disinfectant into the endoscope channels until the channels are filled with the disinfectant, and that no air pockets remain within them. It is absolutely essential that all of the surfaces be in complete contact with the chemical. Soak the endoscope in the high-level disinfectant for the time and temperature required to achieve HLD.
Step 6: Rinsing Rinse with water in accordance with the chemical manufacturer's instructions to remove chemical residue. If rinsing is performed manually, it must include at least three separate rinses with fresh water each time. When rinsing a lumen, it should be flushed with a volume of water that is at least three times the volume of the lumen.
Step 7: Drying Flush lumens with air, followed by 70% isopropyl alcohol until the alcohol can be seen exiting the opposite end of each channel, and followed-up by a second purging of the channels with medical or filtered air to facilitate drying.
Step 8: Inspection There must be an effective quality assurance programme for the settings in which the endoscopes are used, with special emphasis on cleaning and HLD or sterilisation. An effective quality assurance programme is fundamental to the delivery of safe and effective patient care. Elements of the quality assurance programme include supervision, including: Visual inspection of the reprocessing procedure and reprocessed device to identify conditions that can affect cleaning and disinfection effectiveness. Training, including additional training and supervised practice each time a new endoscope model or device is introduced. Training when a cleaning or disinfection product or process is changed. Annual competency review of staff responsible for reprocessing. Records indicating that the manufacturer's recommendations for maintenance schedules of endoscopes are performed.
Step 9: Storage During storage, endoscopes must hang vertically in a well ventilated dedicated area in a manner that minimises contamination or damage. Other validated methods that ensure dry storage must be used. Endoscopes must not be stored in their transport suitcases. Surfaces should be non-porous and cleanable. Storage cabinets must be cleaned at least twice weekly, using a procedure approved by the infection prevention and control department.

Magill forceps

Magill forceps must be steam sterilised after each use. Adequate numbers of Magill forceps per OT should be acquired to allow for this.

Nasopharyngeal and rectal temperature probes

Nasopharyngeal and rectal temperature probes require sterilisation after each use according to manufacturers' recommendations. Adequate numbers of nasopharyngeal temperature probes per OT should be acquired to accommodate this.

Suction bowl

The suction bowl is the container that is filled with water that is used to clear anaesthetic suction catheters or Yankauers™. It should be changed to a plastic or metal receptacle that can be replaced after each patient (one suction bowl per patient). The contaminated receiver should be sent for sterilisation.

Rationale

A study was conducted to determine the prevalence of blood (occult or visible) and/or visible organic material contamination of anaesthetic equipment deemed to be ready for use in theatres in regional, tertiary and central hospitals in KwaZulu-Natal. Of the Magill forceps, nasopharyngeal temperature probes and suction bowls that were examined, 50% (0–100%), 80% (0–100%) and 90% (0–100%), respectively, were contaminated with blood (occult or visible) and/or visible organic material.[6]

Suction bowls are water-filled containers that are used for clearing anaesthetic suction catheters or Yankauers™. Suction bowls do not come into direct contact with the patient's oral mucosa. The suction bowl and water become contaminated with oral secretions, blood or vomitus each time the anaesthetist dips the suction catheter into the water. Changing the water only is ineffective as the bowl becomes contaminated with blood and secretions with each use and will contaminate the clean new water placed in it. Some hospitals also use these bowls as a common receptacle for used laryngeal mask airways and oropharyngeal airways.

A study that examined the decontamination practices of nasopharyngeal temperature probes and Magill forceps in theatres in regional, tertiary and central hospitals in KwaZulu-Natal found that 60% and 53% of the hospitals did not meet the minimum standard required for the reprocessing of nasopharyngeal temperature probes and Magill forceps, respectively.[7]

Sterilisation is recommended as it has a large margin of safety, coupled with reliability, consistency and lethality.[95,115] It also removes the "human" element from the process of decontamination, and is relatively inexpensive.

Suction tubing

Disposable plastic tubing is recommended for suction tubing. The tubing should be replaced after each patient.

Oropharyngeal airways

Single-patient use only is applicable to oropharyngeal airway equipment, which must be discarded after each use.

Bougies, and intubation guides and stylets

A gum-elastic bougie may be disinfected up to five times between patients according to the manufacturer's recommendations. It should be stored in a sealed packet.[9]

Alternative single-use intubation aids are preferable to bougie use. Intubation aids and stylets are single-use items. Rigid stylets for use with video laryngoscopes, e.g. GlideRite with the Glidescope, should be sterilised as per manufacturer's guidelines.[116]

Rationale

Suction tubing connects the suction catheter or Yankauer™ to the suction apparatus. The danger of not changing suction tubing after each patient is that a clean suction catheter may become contaminated once it is attached to the tubing. Once the suction is turned off, secretions, gastric or bowel contents or blood may track down the tubing owing to gravity and contaminate the clean suction catheters or Yankauers™. Oral and nasal airways are single-use items since they readily become contaminated with transmissible organisms and blood.[117]

It has been noted that bougies, and intubation guides and stylets have become contaminated with pathogenic bacteria after reuse, and have been associated with cross-infection.[118,119]

Breathing filters and breathing circuits

Use of a breathing filter must include the following:

- Use a new, high-quality HMEF for every patient. The HMEF must be changed between patients.[120]
- The filter should be placed on the Y-piece between the endotracheal or tracheostomy tube and the elbow connector or breathing circuit.[121-125]
- The high-quality HMEF should be above the level of the lungs, with the filter in a vertical position to decrease the risk of contamination from secretion from the patient or condensate from the breathing circuit.[126]
- The anaesthetist must actively search for complications associated with the use of breathing filters, such as obstruction of the filter with blood or secretions, an increase in airway resistance and possible disconnection.[127-136]
- The filter should not be placed between the circuit and the absorber as this practice can lead to the desiccation of soda lime, with the resultant risk of carbon monoxide poisoning.[120,137-139]
- The filter has to be changed when it becomes visibly contaminated with blood or secretions, or with condensate within the breathing system.[140]

Type of breathing filter

The HMEF must have been tested using the saline test as prescribed in ISO 9360-1:2000 or the European standard norm EN13328-1. The HMEF should have a 99.97% efficiency at a flow rate of 30 l/minute.[141] The HMEF should be able to withstand a pressure of 60 hectopascals (≈ 60 cmH_2O) without allowing liquid to pass through, or 20 hectopascals above the set pressure limit of the breathing circuit.[121,142]

The HMEF must have a minimum humidity output of 20 g/m^3 in patients ventilated < 10 hours or 33 g/m^3 in ICU patients ventilated > 10 hours.[141,143,144] When using low flows, the dead space in the filter should be appropriate for the patient's tidal volume. Ideally, the HMEF should be a hydrophobic pleated filter.[121] Electrostatic filters should not be used in cases where there is a high risk of cross-infection as they do not prevent the passage of liquid through the filter.[121,126,145-150] Electrostatic filters do not prevent transmission because liquid (carrying viruses and bacteria along) can pass through these filters.

The increase in dead space, increased airway resistance and possible delayed inhalational induction of anaesthesia when using breathing filters/HMEFs, should be considered in children. The lower-weight limit should be a HME of 5 kg and filter of 3 kg.[140,151]

Breathing circuits

The breathing system consists of the elbow connector or catheter mount, the breathing circuit, the reservoir bag and CO_2 absorber.

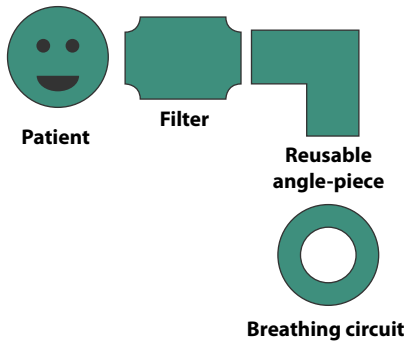

Figure 2: Correct placement of filter

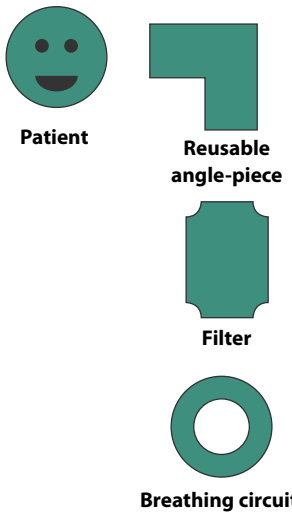

Figure 3: Incorrect placement of filter

The components of the breathing circuit can be reused between cases for up to seven days, provided that:

- A high-efficiency filter has been used.
- There are no defects in the system.
- It has been disinfected according to the manufacturer's instructions daily.
- It has been cleared by the manufacturer to be used as such.[152,153]
- The breathing system components are seen as semi-critical items and should be disinfected according to the manufacturer's instructions.[154]

The CO_2-absorber canister should be cleaned every time the absorber material is changed. Disinfection must take place according to manufacturer's guidelines.[154,155]

The components of the breathing circuit should be changed immediately in any of the following circumstances:[153]

- When it is visibly soiled with blood or secretions.
- When used on a patient with a confirmed or potential notifiable infectious disease that involves the risk of transmission via the breathing circuit and reservoir bag, e.g. tuberculosis, acute viral hepatitis, measles, influenza virus, infection and/or colonisation with a multidrug-resistant pathogen and upper or lower respiratory tract infection.

Oxygen tubing, oxygen masks and nasal prongs

These are single-use items and should be discarded after use on a single patient. An area of at least 0.4 m from the mask should be considered to be a potential hazard for aerosolised pathogens.[108,157] Patients with high-risk respiratory infections should only be nebulised when necessary and should be isolated during nebulisation in a room with good ventilation.

Rationale

The aim of using breathing filters in anaesthetic circuits is to prevent contamination of the breathing circuit and ventilator with pathogens from the patient. An array of pathogens has been found in contaminated circuits. These include: *A. caloaceticus*, *P. maltophilia*, *P. aeuroginosa*, *Flavobacterium meningosepticum*, *K. pneumoniae*, *P. mirabilis*, *Enterobacter cloacae*, *Citrobacter diversus*, *E. agglomerans*, *Candida albicans*, *Proteus* spp. and *Streptococcus* spp.[155,156] It is also to prevent the passage of possibly contaminated liquid out of the circuit back to the patient. In essence, the aim is to prevent cross-contamination. This is particularly important in a country with a high incidence of HIV, tuberculosis and other opportunistic infections. The practice of using viral or bacterial filters allows the reuse of anaesthetic breathing circuits between patients in a cost-constrained environment.

Another benefit of combining a viral or bacterial filter with a HME device is that it prevents the inspissation of secretions, and thus allows for proper airway toilet, therefore decreasing the risk of infection. Dry air also leads to mucosal damage that can become an entry port for microorganisms. Thus, humidification is beneficial in preventing mucosal damage.[141,142,158] Different

devices can be added to the breathing circuit, and it is important for anaesthetists to appreciate the differences between them.

There is a simple breathing filter, without any HME. Usually, this is a yellow filter. Another HME has no filter and is usually blue. While yet another HME has a built-in breathing filter. This is known as an HMEF. These devices are generally green in colour.[141]

Other confusion with the terminology derives from the use of the words "electrostatic" and "pleated". All filters are pleated and have some degree of electrostatic properties. The term "pleated filters" is used to refer to hydrophobic filters.[141,159] The aim of pleating the filter material is to increase the surface area and therefore decrease airway resistance, as well as increasing the filtration area.[140,159]

The efficiency of electrostatic filters decreases over time as the electrostatic charge decreases. It also becomes inefficient when pressurised, so that liquid is able to pass through, and with it, carrying bacteria and viruses. Hartmann et al.[145] demonstrated that up to 5.6% of contamination occurs in breathing circuits that had been in use for 72 hours with electrostatic filters.

All of the above filters have to be subjected to quality control tests by being subjected to the international standard, ISO 9360-1:2000, or the European standard, EN 13328-1.[141,160] This requires the filters to be challenged with particles in the order of 0.3 µm. This size particle is the size that is most likely to be able to penetrate a filter. Particles below this size are captured by Brownian diffusion, while those that are larger are trapped by inertial impaction and interception.[141] Efficiency is measured by the percentage of particles that get trapped by the filter. Typically, this ranges from 95–99.97%. Thus, efficiency is always reported as a percentage. Other symbols commonly found in the terminology are the letters "N", "P" or "R" in front of the efficiency percentage, e.g. N95. This refers to the ability of the mask to withstand oil. "N" means not oil proof, "R" means resistant to oil and "P" means oil proof. The "N" masks are the most commonly used masks in the medical field.

There has been a longstanding debate on whether or not a new circuit should be used for every patient, or whether a filter can be used to avoid contamination and then the circuits reused among patients. Some studies have shown that filters placed at the Y-connection were able to prevent the contamination of breathing systems.[122-125] It is also known that filters reduce airborne microbes when tested in vitro.[146-148,161,162] Evidence by Von Hassel showed that the risk of acquiring a lower respiratory tract infection from shared breathing circuits without using filters was very low at 0.1–0.2%. He subjected the circuits to HLD and pasteurisation and only used filters in HIV-reactive patients and those with tuberculosis. However, low-flow anaesthesia was not used, and thus, it is possible that the high gas flows had washed any contaminants out of the circuit. The American Thoracic Society[164] noted that although HMEFs reduce circuit colonisation, they do not significantly reduce the incidence of ventilator-associated pneumonia and cannot be regarded as a tool to be used to totally prevent ventilator-associated pneumonia.

What is clear is the risk of complications associated with the use of filters.[127-139] There have been case reports of patients suffering fatal events when breathing filters became blocked with blood or secretions. This led to an increase in airway resistance and could have contributed to an increase in dead space. HMEFs may also be associated with a risk of carbon monoxide poisoning. In particular, this is problematic when the HMEF is placed between the circuit and the absorber. This can lead to the desiccation of soda lime or bara lime, with the resultant production of carbon monoxide. Carbon monoxide poisoning might not be easily detected as the oxygen saturation monitor will give a false reassuringly high reading. The risk is also higher when higher gas flows are used.

This has led to North American societies recommending the single use of a breathing circuit per patient without the use of an airway filter. European societies have taken a different stance in view of cost constraints, and in their guidelines have recommended that breathing circuits can be reused, as long as a high-efficiency HMEF is utilised at the Y-piece and changed between patients, or when it becomes visibly soiled. In addition, the filters have to be able to withstand a pressure of 60 hectopascals in order to prevent liquid from condensation within the circuit passing back through the filter, carrying bacteria and viruses with it.[121,142] The British Society for Antimicrobial Chemotherapy[120] recommends the use of HMEs rather than heated humidifiers to reduce the incidence of ventilator-associated pneumonia.

When circuits are to be reused, it is recommended that a high efficiency filter is used. Chant et al.[165] reported on the possible patient-to-patient transmission of hepatitis C. A case series was described in which five of the patients on the elective minor operations list developed hepatitis C approximately seven weeks after surgery. All of the viruses where typed to be the same strain. A second such case was also reported.[166]

Various pathogens are, or have, the potential to be airborne over long distances (> 1 m). These pathogens pose a high risk of contamination of the breathing surface and ventilator, as well as the surrounding environment. Tuberculosis and influenza viruses are examples of such pathogens.

When reusing circuits between patients, it must be ensured that the circuit is classified as such by the manufacturer. Reusing circuits that are classified as "single-use" makes the anaesthetist liable.[167] The manufacturers of multi-use circuits have specific instructions on the disinfection and cleaning of such circuits, and these have to be followed carefully to ensure continued performance.

Masks and tubing used for oxygen therapy are contaminated with secretions from the patient's upper airways. They should never be reused between patients. Ip et al.[156] demonstrated that aerosolised droplets could be detected as far as 0.4 m from a simple face mask during normal breathing. This distance can increase dramatically when the patient sneezes, coughs or vocalises. Patients with high-risk infections should only be nebulised when necessary, and in rooms that are isolated and

have good ventilation. The risk is that nebulisation increases the velocity and distance in which aerosolised particles can travel. Nebulisation was thought to be the main cause that led to an outbreak of severe acute respiratory syndrome in Hong Kong in 2003.[168]

Bag valve mask resuscitators

All resuscitators should be fitted with a high-efficiency breathing filter between the valve and the mask before being used on a patient. Resuscitators used on the same patient should be capped at the patient connection port when not in use. Resuscitators should be cleaned and disinfected according to the manufacturers' instructions. The resuscitator should be disassembled and all the parts washed thoroughly, using clean water and mild detergent. It is necessary to ensure that the detergent is suitable for the material. Do not disassemble the pressure release valve and the positive end expiratory pressure valve. All of the parts should be rinsed in clean water to remove the detergent. All of the parts should be allowed to dry in a clean controlled environment, where the risk for recontamination is low. The components should then be subjected to one of the following decontamination techniques:[14]

- Pasteurisation for 30 minutes (not the oxygen reservoir bag).
- Autoclaving not to exceed 132 °C (not the oxygen reservoir bag).
- Ethylene oxide gas (all parts are suitable).
- Liquid sterilisation (all parts are suitable) with Cidex® OPA or sodium hypochlorite. Wash thoroughly to remove any excess disinfectant.

Manual resuscitator should be sterilised:

- For first-time use.
- Between patients.
- When visibly contaminated.
- Every 24 hours of use in the same patient.

Rationale

Manual resuscitators are classified as semi-critical items and should be decontaminated using HLD or sterilisation. It is important to follow the instructions of manufacturers in this regard as certain parts of the resuscitator might not be compatible with the chosen method of sterilisation. It has been shown that bacteria can be cultured from resuscitators which were macroscopically "clean". Paediatric Ambu® bags in the obstetric OT are used more frequently than adult ones and have been linked with disease transmission.

Supraglottic devices

Single-use (disposable) SADs are preferred to reusable SADs.

If reusable SADs, e.g. LMA Classic™, are used, they should be sterilised in an audited sterile service department and not more often than recommended by the manufacturer, e.g. 40 times for LMA Classic™. Do not decontaminate and reuse single-use SADs.

Rationale

Reasons for the preferred use of disposable SADs as opposed to reusable SADs include:

- *The potential for the iatrogenic spread of protein-born prion diseases:* The main concern in the reuse of the laryngeal mask airway (LMA) is the potential for the iatrogenic spread of protein-born prion diseases. Prions are proteins which become distinctive infectious agents that can reproduce without nucleic acids, and can cause transmissible spongiform encephalopathies in humans, including Kuru and Creutzfeldt–Jakob disease, under certain conditions.[169] Variant Creutzfeldt–Jakob disease, a transmissible prion disease, was identified in humans in Great Britain in 1996.[170] Recently, large numbers of prion proteins were detected in human tonsillar tissue, raising significant concern in several countries about reusable anaesthetic and surgical equipment.[171] Concerns were raised in Europe about the SAD owing to its close proximity to tonsillar tissue. Prions are extremely resistant to inactivation by disinfectant chemicals and heat. Accordingly, the cleaning and autoclaving of reusable SADs may not be adequate to prevent the iatrogenic spread of prions. Several studies have found significant protein contamination on reusable SADs, even after cleaning and autoclaving.[117,172-174] The risk to patients is as yet unquantified owing to several challenges, such as the long incubation period between exposure to prions and the development of clinical features, combined with an unknown number of carriers, undetermined exposure times and incomplete penetration. However, investigators have estimated that the number of carriers of variant Creutzfeldt–Jakob disease could range from thousands, to as many as millions of people worldwide.[175] Moreover, the risk to anaesthesia staff during both the use and decontamination of the LMA has not been considered.
- *Monitoring of reuse:* Tracking the number of times that the reusable SAD is reused is essential. However, this is often not carried out. The original LMAs were designed for use up to 40 times.
- *Affordability:* Studies have shown that the cost of a disposable SAD compares favourably with the cost per use of a reusable SAD, even when staff time and cleaning costs were excluded.[176]
- *Efficiency:* There is no need to allocate time to the cleaning and sterilisation of the LMAs.
- When surveyed, the majority of clinicians would want single-use devices to be used on their families and themselves if they were patients.[11]

Storage of semi-critical items

Semi-critical items should be packaged and stored in a way that prevents recontamination. Suggested compliant storage methods include a peel pouch or a closed plastic bag. These items should not be left unwrapped in or on top of anaesthesia workstations and trolleys.

Non-critical medical equipment surfaces

Non-critical medical equipment surfaces extend to blood pressure cuffs, stethoscopes, and frequently used control mechanisms, e.g. pop-off knobs, flow controls and vaporisers. It is necessary to disinfect with a low- or intermediate-level disinfectant after each patient. Medical equipment surfaces can become contaminated with blood and infectious agents and contribute to the spread of healthcare-associated infection.[177,178] Loftus et al.[179] recently reported that multidrug-resistant bacterial transmission to the anaesthesia work area occurred during the practice of general anaesthesia.

Disposable devices

"It seems to me that reusing disposable devices has an element of poetic justice ingrained, if one can become poetic about economics."
—VW Greene

A single-use medical device is to be used on a single patient during one procedure.[113] The reuse of disposable or single use devices started in the 1970s and is a growing and common practice worldwide, especially in resource-limited settings.[113,180-182] Informed consent should be obtained from the patients if an item is to be reused on them. Certain issues exist, including the decontamination technique and the risk of cross-contamination, material alteration, a clear limit to the number of times that an item can be reused, mechanical failure of the device, exposure risks to health care workers and ethical and legal implications.[80,183] Although the reuse of single-use devices is strongly discouraged, the suggested practice from the ISID[80] and the WHO[184] are as follows:

- A facility should be committed to the reuse of single-use devices and have an institution-specific policy with clear guidelines.
- The disposable devices should be classified and reprocessed as per their intrinsic risk: critical, semi-critical and non-critical.
- Functionality and integrity of the device should be maintained.
- The package labelling and manufacturer's guidelines should be followed.
- Reprocessing of disposable devices should be cost justified and performed by a licensed reprocessor.
- Both the physician and the patient should be informed that a device being used is a reprocessed single-use device.
- Any person who reuses a single use device takes full responsibility for its safety and effectiveness.
- A platform for reporting any adverse events from the reuse of single-use devices should be available. The Association of Perioperative Registered Nurses (AORN) has recommended that the sterility, integrity and functionality of a reprocessed single-use device must be documented as safe for patient care and/or equal to the original device specifications.

Trans-oesophageal echocardiogram probes (TEE)

A semi-critical device with increasing usage in anaesthetic practice that has the potential for cross-contamination, especially if the probes are damaged.[12] A sheath should preferentially be used but does not eliminate the risk of contamination and does not exclude the probe from HLD. The recommended basic principles for reprocessing TEE probes are:[12]

- Clean the probe shaft and tip either with immersion or with a detergent-moistened wipe or enzymatic cleaner to remove gross contamination.
- Use a second wipe to wipe the proximal non-immersible parts such as the handles.
- Ensure that there is no structural damage to the probe.
- Use HLD to disinfect the probe tip and flexible shaft.
- Thoroughly rinse and dry before storage.
- The manufacturers' instructions regarding chemical disinfectants should be followed.

Chapter 7: Prevention of central venous line-associated infection

As anaesthesiologists are frequently responsible for the placement of invasive lines in both the theatre and ICU environment, sound guidance with respect to this process will follow. These guidelines particularly focus on interventions which reduce the incidence of infections and sepsis associated with these devices.

Minimisation of catheter use, or alternatives are important aspects of preventing catheter-related bloodstream infections (CRBSIs). Anaesthetists should identify evidence-based indications for the insertion of central venous catheters.[185]

Evidence that simple interventions reduce the incidence of sepsis significantly is key to the implementation of these guidelines.[185] Institutions should be able to produce data on the incidence of CRBSIs and promote care bundles and interventions that have been shown to reduce the incidence of CRBSIs effectively.

The placement and securing of central venous catheters

- The subclavian site is preferred over either the internal jugular or femoral sites in adult patients in order to reduce the incidence of sepsis and of thrombosis.[186-188]
- If the patient has chronic kidney disease, consider the internal jugular vein to avoid subclavian vein stenosis.[189]
- In children and infants there is no preferred venous site to minimise the risk of infection.
- Use ultrasound when possible and when trained operators are available to reduce the time to cannulation, the number of cannulation attempts and the incidence of mechanical complications.[190-193]
- Use a line with the minimum number of lumens necessary to facilitate management of the patient.[194-197]

- When adherence to sterile technique cannot be assured, the line must be removed as soon as possible, but within 48 hours.[198]
- An engineered stabilisation device that is designed specifically to control movement at the catheter hub is recommended, as sutures increase the risk of infection. Standard dressings and tape are not suitable alternatives.[198]
- All lines that are no longer needed should be removed promptly.

Rationale

The optimal insertion site depends on the expected duration of catheterisation and on the type of complication that is most detrimental for the patient overall. The subclavian site has a lower risk of catheter-related bloodstream infections, and a lower thrombotic risk, in comparison to internal jugular and femoral venous sites, particularly for dwell times exceeding five days. The femoral route for central venous access is prone to both infection and thrombosis. The subclavian site is on a flat, relatively immobile portion of the anatomy and is infrequently contaminated with fluids such as saliva, vomit or urine. In addition, the site is usually not subject to the accumulation of sweat. It is important to note that in one study, the subclavian venous site was associated with mechanical complications in 17% of patients when compared to the femoral site.[186,187]

Ultrasound, in the hands of individuals who have been appropriately trained, has been shown to be effective in reducing time to cannulation, the number of attempts, and the mechanical complication rate, whilst increasing the rate of procedural success.

Limited data exist regarding the risk of CLABSI with single- versus multi-lumen central venous catheters. Studies have demonstrated increased infections in various patient populations in whom multi-lumen catheters are present, although a meta-analysis did not find a significant difference.[196] Guidelines tend to recommend using catheters with the minimum number of ports that are essential for patient management.

Non-adherence to sterile technique should not be condoned, and lines must not be placed in such circumstances unless immediately life-saving. Central venous catheters must be secured distally to prevent the loss of venous access and to reduce movement at the insertion site, which increases the risk of bloodstream infections. Sutures are associated with an increased risk of infection as they support the growth of biofilm. Specifically-engineered stabilisation devices are recommended. Standard dressings and tape are not suitable alternatives. Lines that have been removed carry no risk of sepsis whatsoever and forms the first step in managing catheter-related bloodstream infections.

Sterile technique for the placement of central venous catheters

- The operator should scrub, as for a surgical procedure, prior to the placement of a central venous catheter.[197-201]
- Maximal sterile barrier precautions to be used include the use of a cap, mask, sterile gown, sterile gloves and a sterile full body drape.[197-201]
- For skin decontamination prior to catheter insertion, the application of sterile > 0.5% chlorhexidine gluconate in 70% isopropyl alcohol represents standard of care.[194] If there is a contraindication to the use of chlorhexidine, tincture of iodine, an iodophore or 70% alcohol may be used as alternatives.[202-204]
- No comparison has been made between using chlorhexidine preparations with alcohol and povidone-iodine in alcohol to prepare clean skin.[202]
- No recommendation can be made for the safety or efficacy of chlorhexidine in infants aged < 2 months.
- The skin antiseptic must be allowed to dry (at least 30 s) prior to performance of the procedure.[197,198]

Rationale

The use of maximal barrier precautions and a sterile technique have been shown to reduce the incidence of line sepsis. Chlorhexidine-containing solutions appear to be optimal with respect to the reduction of septic complications. It is, however, associated with more cutaneous skin reactions. Severe allergy and anaphylaxis to chlorhexidine, though rare, has been reported. Application technique, correct dose for the skin surface area and drying time (at least 30 s) are important components of adequate skin decontamination. A lower alcohol concentration may improve the skin permeation of the antiseptic

Catheter dressing and site management

- Do not use topical antibiotic ointment or creams on insertion sites as they have the potential to promote fungal infections as well as antimicrobial resistance.[197]
- A sterile, transparent semi-permeable polyurethane dressing must be used to cover the site. Sterile gauze and tape may be used as an alternative.[197,198,205,206]
- Sterile gauze should be used if there is any bleeding, exudate or excessive skin moisture that accumulates around the insertion site.[198]
- The dressing must be replaced immediately if there is any sign that it is becoming loose, if there is any soiling, or if any dampness is noted under the dressing or at the insertion site. When replacing the dressing, the skin must be cleaned with antiseptic and allowed to dry before applying the new dressing.[198]
- Gauze dressings must be replaced at least every two days.[197,198]
- Clear transparent dressings must be replaced at least every seven days.[197,198]
- Line sites must be monitored daily especially at the catheter-skin junction site and surrounding area for pain, erythema, swelling or purulence, which may indicate infection, phlebitis, infiltration, or catheter-associated venous thrombosis.[198]

Rationale

Semi-permeable dressings allow for the evaporation of moisture from below the dressing. Keeping an insertion site dry and

avoiding maceration of the surrounding tissue is of paramount importance in preventing infection. Regular monitoring of the line site leads to early detection of sepsis and thrombosis.

The use of catheters and dressings that have associated antimicrobial activity

Impregnated sponge-type dressings are only recommended if the CLABSI rate is not decreasing despite adherence to basic prevention measures, including education and training. This includes adherence to all of the previously described principles.[207-209] Antimicrobial-impregnated catheters or those with antimicrobial properties may be considered in environments in which the rate of CLABSI is not decreasing, and if lines are likely to remain in place for more than five days.[197]

Rationale

The basic principles of infection control and line management previously described cannot be substituted with the use of catheters or dressings with antimicrobial activity.

The management of lines and administration sets

- In patients not receiving blood, blood products or fat emulsions, replace administration sets that are continuously used, including secondary sets and add-on devices, no more frequently than at 96-hour intervals, but at least every seven days.[209-211]
- Infusion sets that have contained blood or blood products or fat-containing emulsions must be changed within 24 hours of initiating the infusion.[210-212]
- Tubing used to administer sedative drugs with a fat emulsion-based vehicle should be changed when the vial is changed, or at least every 12 hours.[212,213]
- When using needleless connectors, use a luer-locking mechanism to ensure a secure connection to the central venous catheter hub or access site.[198]
- When needleless systems are used, a split septum valve may be preferred over some mechanical valves due to increased risk of infection with the mechanical valves.
- Disinfect needleless connectors prior to each entry into the device.[197,198]
- Use aseptic no-touch technique to change the needleless connector.[198]
- Change the needleless components at least as frequently as the administration set. There is no benefit to changing these more frequently than every 72 hours.[214,215]
- Needleless connectors and access ports on administration sets must be cleaned with 70% alcohol, tincture of iodine or chlorhexidine, prior to injection or connection.[198]
- Access needleless connectors only with a sterile device. [197]

Rationale

The infusion of blood or blood products or fat-containing emulsions increases the risk of infection. The need for a needleless connector placed between the catheter hub and the administration set used for continuous fluid infusion is unknown. The main role of needleless connectors is to protect healthcare personnel by eliminating needles and needlestick injuries when attaching administration sets and/or syringes to the catheter hub or injection site. Unless strict cleaning procedures are adhered to, the use of needleless connectors is no safer from an infection perspective.

The insertion of peripheral catheters

- Strict hand hygiene must be observed before and after the placement, removal or palpation of the catheter-insertion site.[43,216,217]
- Strict hand hygiene is also required before and after accessing or dressing a catheter.[43,216,217]
- Clean gloves, rather than sterile gloves, should be used for the insertion of peripheral venous catheters if the access site is not touched after the application of skin antiseptics.[198]
- Sterile gloves should be used for the insertion of arterial and umbilical catheters.[198]
- Clean gloves must be worn when changing the dressings on intravascular catheters.[198]
- Skin preparation with 70% alcohol, tincture of iodine or chlorhexidine is acceptable for peripheral venous catheter insertion.[218,219]
- A cleaning solution that contains more than 0.5% chlorhexidine in alcohol should be used when inserting arterial catheters.[218,219]
- The upper extremity should ideally be used for the insertion of venous catheters in adults.[198,219]
- The upper or lower extremities, or the scalp, may be used as a catheter insertion site in paediatric patients.[198]
- The radial or dorsalis pedis site is preferred over the femoral or axillary site to reduce the risk of infection in adults.[220,221]
- Remove the peripheral catheters if there is any sign of infection or inflammation at the insertion site. This includes redness, tenderness, purulence or obvious thrombophlebitis.[218,219]
- While many of these recommendations may already be in practice, practitioners should be careful to take heed of statements that are made. The risks, incidence, precipitating factors and preventive strategies are all well researched and subject to a wealth of evidence.
- Extensive reviews and recommendations are published by the Centers for Disease Control and Prevention (CDC) and Healthcare Infection Control Practices Advisory Committee.[222,223]

Chapter 8: Infection control recommendations for regional anaesthesia

Spinal, epidural and caudal procedures

A historical and physical examination and review of relevant laboratory studies should be conducted to identify patients who may be at risk of infectious complications. These include patients with retroviral disease, uncontrolled diabetes, oncology patients, post-transplant, etc. Consider alternatives to neuraxial techniques for patients who are at high risk.

When a neuraxial technique is used in a patient with known or suspected bacteraemia, prophylactic pre-procedure antibiotic therapy should be considered.

Aseptic technique should always be used during the preparation of equipment, e.g. ultrasound, the preparation of drugs to be administered, and the placement of neuraxial needles and catheters.

A caudal anaesthetic is considered to be a neuraxial technique as the caudal space is a continuation of the epidural space.

Maximal barrier precautions apply:

- Jewellery should be removed, and hands washed.
- Caps, masks (covering both mouth and nose and all facial hair), sterile gloves and gowns should be worn.
- Sterile drapes should be used.
- A face mask should also be worn by the anaesthetic assistant.
- An antiseptic, preferably a 0.5% solution of chlorhexidine with 70% alcohol, should be used for skin preparation, and adequate time allowed for drying.
- Meticulous care should be taken to avoid contact of chlorhexidine with the cerebrospinal fluid.
- Do not pour chlorhexidine into containers in close proximity to equipment that will be used for the neuraxial anaesthetic. Cover or protect equipment while cleaning with chlorhexidine.
- A sterile occlusive dressing must be applied over the catheter insertion site.
- Bacterial filters may be considered during extended continuous epidural infusion.
- Disconnection and reconnection of the neuraxial delivery system should be limited.
- The removal of unwitnessed, accidentally disconnected catheters should be considered.
- Catheters must not remain in situ for longer than clinically necessary.

Rationale

Infectious complications of neuraxial anaesthesia include meningitis, encephalitis and epidural abscesses. The frequency of meningitis is estimated to range from 0.2–1.3 per 10 000, and that of epidural abscess to be one in 145 000.[224] Although rare, the consequences of these complications are disastrous. In the *Saving Mothers 2008-2010: fifth report on the Confidential Enquiries Into Maternal Deaths in South Africa*, one maternal death was attributed to post-spinal meningitis.[225]

Meningitis usually occurs within 6–36 hours after dural puncture. It can be mistaken for post-dural puncture headache. The latter should always be a diagnosis of exclusion. Meningitis is usually associated with fever, neck stiffness, photophobia and increased septic markers. Prompt treatment with antimicrobial drugs is indicated as a delay can be associated with worse outcomes.

Chlorhexidine and alcohol are toxic to the tissues, and care should be taken not to contaminate the needles. Always allow alcohol to evaporate before inserting the needle.

The recommendations are mainly based on a practice advisory produced by the American Society of Anesthesiologists (ASA) in 2010.[226] Guidelines produced by the Association of Anaesthetists of Great Britain and Ireland (AAGBI)[9] and the Australian and New Zealand College of Anaesthetists (ANZCA) make similar recommendations.[227]

As infectious complications associated with neuraxial procedures are uncommon, evidence from the literature on the efficacy of interventions in their reduction is also scarce. The result is that the literature is insufficient in determining whether or not any of the interventions reduce infectious complications associated with neuraxial techniques.[226]

Patient factors

No controlled trials have addressed the impact of a focused history, physical examination or laboratory evaluation on infectious complications, but several studies suggest certain patient characteristics, e.g. cancer, diabetes and an impaired immune response, may be associated with neuraxial infection.[226]

Jewellery

Watches and rings should be removed during handwashing.[228,229] Higher microbial counts have been found in healthcare workers who do not remove rings during handwashing.[229]

Gowns

ASA members and consultants were equivocal with regards to the wearing of gowns.[226] The AAGBI includes gowns as part of its maximal barrier precautions, which it recommends for neuraxial techniques[9] There are insufficient data to make definitive recommendations with regards to routine gown use in this context.

Surgical face masks

This area is also not without controversy. However, in cases of neuraxial-associated meningitis in which the pathogen was known, 24% of the infections were of skin origin, while an overwhelming 76% were attributed to mouth commensals.[230] Furthermore, Schneeberger reported on a cluster of four

patients who developed streptococcal meningitis after spinal anaesthesia, performed by the same anaesthesiologist who was under treatment for recurrent tonsillitis and who did not wear a mask.[231] North and Brophy described an epidural abscess that was proven to be caused by a strain of *Staphylococcus* cultured from the nose of the anaesthetist who placed the epidural catheter.[232] In addition, Philips et al. showed that wearing a face mask resulted in marked reduction in the bacterial contamination of a surface in close proximity (30 cm) to the upper airway.[233] Therefore, in line with a recent CDC recommendation, the use of a surgical face mask is recommended when performing a neuraxial procedure.[234]

Skin preparation

Two randomised controlled trials indicated that the rate of positive bacteriological cultures was reduced when the patient's skin was prepared with chlorhexidine, rather than with povidone-iodine, before epidural catheterisation.[235,236] The anaesthetist must exercise extreme caution to prevent the contact of chlorhexidine and alcohol with cerebrospinal fluid and neural structures as both are neurotoxic. The 0.5% solution is preferred over the 2% solution due to the lack of clear evidence for the use of the one over the other. The antimicrobial action of alcohol is denaturation of proteins and its dehydrating properties. Therefore, it must be allowed to dry completely before the procedure.

Bacterial filters

Three observational studies showed that infections and epidural abscesses occur in the presence of micropore filters.[237-239] Therefore, an aseptic technique should be used when drawing up drugs to be administered via the neuraxial route.

Duration of epidural catheterisation

Observational studies indicate that infections and epidural abscesses occur in the presence of longer duration of catheterisation.[240-244]

Peripheral nerve blocks

- Maximal barrier precautions are generally not necessary. Maximum barrier precautions should be used if the patient is immunocompromised or a perineural catheter needs to be inserted.
- Jewellery should be removed, and hands washed. Sterile gloves must be worn. Aseptic techniques should always be used during the preparation of equipment, e.g. ultrasound, the drawing up of drugs and the placement of needles and catheters.
- An antiseptic, preferably chlorhexidine with alcohol should be used for skin preparation and adequate time allowed for drying.
- Do not put the probe on the patient until the alcohol has evaporated to prevent alcohol induced damage to the probe.

The insertion of a perineural catheter

- Use a sterile occlusive dressing.
- Limit disconnection and reconnection of the delivery system.
- Catheter should not remain in situ for longer than is clinically necessary.

The use of ultrasound

A sterile probe and handle covering should be used, e.g. a sterile transducer sheath. The ultrasound machine and probe should be decontaminated before and after use by following the instructions from the manufacturer manual. Product information should be consulted with regards to which cleaning agents are appropriate for the specific machine or probe. Use single-use, sterile gel, e.g. K-Y® lubricating gel sachet.

Rationale

Even with a perineural catheter in situ, infectious complications relating to peripheral nerve blocks are rare, e.g. local infections of 0–3.2% and abscess formation of 0–0.9%.[245] The risk is much smaller still for "single-shot" blocks. There have been only occasional case reports in the literature.[246]

Unless a perineural catheter is to be placed, or the patient is immunocompromised, both the AAGBI and ANZCA guidelines agree that maximal barrier precautions are unnecessary when performing peripheral nerve blocks.[9,227] If a perineural catheter is to be inserted, the procedure should be as for the previously described neuraxial techniques.

Ultrasound transducers and gel have both been implicated in outbreaks of hospital-acquired infection.[247-251] There are no universally adopted guidelines for the decontamination of ultrasound machines and transducers used for ultrasound-guided regional anaesthesia. The result is a wide variety of different practices. Only gel that is manufactured to be used as ultrasound gel should be used. There are a number of substances in the various gels that can cause physical damage to the probe. Such substances include alcohol-based solutions, mineral oil, olive oil, iodine, lotions, aloe vera and methyl/ethyl parabens.

The Spaulding classification recommends different levels of decontamination of medical equipment, depending on the risk of infection. It categorises equipment as critical (a high risk of infection), semi-critical (equipment in contact with mucous membranes or non-intact skin) and non-critical (equipment in contact with intact skin).[95]

Ultrasound-guided regional techniques involve skin puncture and possible blood contamination of the transducer. Therefore, in this context, ultrasound probes for regional procedures should be considered to be semi-critical equipment, which, according to the Spaulding classification, requires full sterilisation or the use of a sterile sheath, followed by prolonged immersion with high-level disinfectants. It is however of critical importance to consult with the manufacturers to ensure that the correct methods are used to prevent damage of the ultrasound probes. The ultrasound probe needs to be cleaned after every use by following the user manual.

Washing the ultrasound probe

- Remove coupling gel with a soft cloth.
- Wash the probe with mild soap in lukewarm water.
- A soft cloth or gauze should be used to remove all visible soap. Air dry or dry using a clean, soft cloth.
- Refer to the user manual to determine the immersion level of each specific probe.

Disinfection

- Please refer to the user manual.
- For disinfection to be effective, all visible contamination should be washed off.
- Liquid germicides such as 2% glutaraldehyde-based solutions are used to disinfect the probe.
- Cidex® is the only liquid disinfectant that has been evaluated for compatibility with the materials of the probe.
- The rinsed and dried probe should be placed in the Cidex® solution for the time specified by the germicide manufacturer.
- Following this, the probe should be thoroughly rinsed to remove any residual disinfectant.
- The transducer can be damaged by contact with alcohol-containing solutions, ammonium chloride compounds, hydrogen peroxide and temperatures above 60 °C

Enforcing this level of decontamination would impose an extra burden on theatre staff and reduce the availability of equipment. It is also unclear whether or not this level of decontamination is required for ultrasound-guided regional anaesthesia, or if a modification is acceptable.[252]

Ideally, a sterile sheath should be used for all peripheral nerve blocks especially in the following cases:[253]

- Patients with multidrug-resistant organisms.
- Patients undergoing perineural catheter placement.
- When there is a risk of blood contamination of the probe.

Taking into account Spaulding's classification, South Africa's context and the burden of disease, and the fact that blood contamination of the probe may be unpredictable, use of a sterile sheath that covers both the probe and the handle is recommended. (Large size sterile gloves have been used in some centres where sterile sheaths were not available.)

In 2004, Health Canada issued a warning about the risk of infection from ultrasound gel and recommended that sterile gel is used for invasive procedures, including needle localisation.[254] In 2012, Oleszkowicz et al.[255] proposed similar guidelines for the USA. In line with these, the use of sterile, single-use, ultrasound gel for invasive procedures is recommended.

Chapter 9: Antimicrobial prophylaxis for surgical procedures

Altemeier classification of surgical wounds[256]

Class 1: Clean SSI risk < 1%	Sterile area of body. Skin intact before surgical incision. Surgery does not involve opening of gastrointestinal, respiratory, genito-urinary or oropharyngeal tracts.
Class II: Clean contaminated SSI risk 2–5%	Opening of body cavities; gastrointestinal tract, respiratory tract, genito-urinary tract or oro-pharyngeal tracts in the absence of gross contamination.
Class III: Contaminated SSI risk 5–10%	Massive surgical soiling by gastrointestinal contents or opening of genito-urinary or biliary tracks in patients with tract infections. Recent open traumatic wounds.
Class IV: Dirty SSI risk > 10%	Body site that contains pus, foreign body or faeces. Traumatic open wounds > 4 hours.

SSI – Surgical site infection

There are four considerations to keep in mind when prescribing antibiotic prophylaxis:[257]

1. Who needs antibiotic prophylaxis?
2. What are the factors that influence antibiotic choice?
3. Timing.
4. Duration of antibiotic prophylaxis.

Procedures that need antibiotic prophylaxis

- Class I surgery involving the placement of prosthesis or implants.
- Class II surgery.
- Class III and IV should be on curative antibiotics.

In procedures where antibiotic prophylaxis would not normally be used, it should be considered in patients with an increased risk for surgical site infections (SSIs) or where infection will be catastrophic. Patients at increased risk for infection include geriatric and oncology patients, those with diabetes, HIV, obesity, transplant patients, etc.[258]

Do not give antibiotic prophylaxis for clean, non-prosthetic uncomplicated surgery.[257,259]

Choice of prophylactic antibiotics

Use an antibiotic that is safe, inexpensive and a bactericidal with an in vitro spectrum that covers the most probable intraoperative contaminants for the operation. Other factors that may influence the choice of antibiotics include renal function, other comorbidities, recent antimicrobial use, institutionalised patients, known colonisation with a drug-resistant organism as well as impaired immunocompetency.

Use your local antibiotic formulary, and always consider potential adverse effects when giving antibiotics for prophylaxis. For a detailed table of surgical procedure and the best choice of prophylactic antibiotics, see Appendix A.[257]

Timing of prophylactic antibiotics

Give a single dose of antibiotic prophylaxis intravenously 30 minutes before skin incision. Timing of administration should be no longer than one hour before incision. Antibiotics that need to be administered as infusions should ideally be completed 30 minutes prior to skin incision. Consider earlier administration of antibiotic prophylaxis for operations in which a tourniquet is used.

With regards to Caesarean delivery, antibiotic prophylaxis should not be delayed to the clamping of the cord.[258] A second dose of an antibiotic with a relatively short half-life, e.g. cephazolin, is often recommended for prolonged procedures or procedures with massive blood loss.

Duration of prophylactic antibiotic therapy

Antibiotic prophylaxis should be brief and limited to the surgical time. It is sometimes used up to 24 hours and under very exceptional circumstances for up to 48 hours. Prophylaxis should never continue for more than 48 hours.

Rationale

- Antibiotic prophylaxis reduces bacterial inoculum at the time of surgery. It significantly decreases the rate of bacterial contamination of the surgical site.
- Antibiotics have associated risks. Adverse effects include gastrointestinal symptoms (nausea, vomiting or diarrhoea), antibiotic-associated *Clostridium difficile* and minor allergic reactions, such as skin rashes, myalgias and arthralgias. Rare adverse effects may include pancytopenia, renal dysfunction, liver dysfunction and life-threatening anaphylaxis.
- Hospitals should develop local guidelines for surgical antibiotic prophylaxis, based on local infective microbes and their antibiotic resistance patterns. They can be formulated by local microbiologists, in consultation with surgical colleagues, and must then be adhered to in the perioperative setting.
- In most clean wounds, SSIs would be due to skin flora. The most common pathogens are skin flora microbes, especially the *Streptococcus* spp. and *Staphylococcus* spp. First-generation cephalosporin, e.g. cefazolin, adequately covers these organisms in a cost-effective manner.[260] Surgeries that involve the bowel necessitate Gram-negative and anaerobic coverage, for which cefoxitin is appropriate. Use of broader spectrum antimicrobial drugs such as co-amoxiclav may be considered based on cost, safety profile and ease of administration (when metronidazole is unavailable). Vancomycin may be the prophylaxis of choice when a "cluster of methicillin-resistant *S. aureus* mediastinitis, or incisional SSI owing to methicillin-resistant, coagulase-negative staphylococci, has been detected".[258,260]
- Studies have shown that the minimum inhibitory concentration of the antibiotic agent at tissue level must be exceeded for the period from incision to wound closure. Thus, the timing of the prophylactic antibiotics is crucial.
- Infection rates are lowest if antibiotics are administered within 30 minutes of incision, with the odds of infection increasing twofold if antibiotics are administered after incision, or 60 minutes before incision. If a fluoroquinolone is used, consider starting the dose 120 minutes before skin incision.[258]
- The WHO initiative, "Safe surgery saves lives" surgical safety checklist emphasises the inclusion of antibiotic prophylaxis given 60 minutes before skin incision.[261]
- Always consider the timing and pharmacokinetics, for example, the serum half-life, and necessary infusion time of the antibiotic (vancomycin). Repeat the dose of antibiotic when the operation is longer than the half-life of the given antibiotic or if there is excessive blood loss. Note that the dose should be adjusted for patients with delayed clearance of the drug.[258,262]

See Appendix A for a complete summary of antibiotics for specific surgical procedures.[257]

APPENDIX A: Antibioprophylaxis in surgery and interventional medicine (adult patients). Update 2017[257]

These guidelines are based on guidelines released by the French Society of Anesthesia and Intensive Care Medicine (SFAR). Local practices may differ from these guidelines.

Table I: Neurosurgery

SURGERY	PRODUCT	INITIAL DOSE	RE-INJECTION DOSE AND DURATION
CSF shunt	Cefazolin	2 g slow IVI	Single dose 1 g IVI if surgery > 4 h
	If allergic: Vancomycin[a]	30 mg/kg slowly IVI over 120 min	Single dose
External CSF shunt	No antibiotic prophylaxis		
Craniotomy	Cefazolin	2 g slow IVI	Single dose (if surgery > 4 h re-inject 1 g)
	If allergic: Vancomycin	30 mg/kg slowly IVI over 120 min	Single dose
Neurosurgery transsphenoidal routes and trans-labyrinthine	Cefazolin	2 g slow IVI	Single dose 1 g IVI if surgery > 4 h
	If allergic: Vancomycin	30 mg/kg slowly IVI over 120 min	Single dose
Spine surgery with implantation of prosthetic material	Cefazolin	2 g slow IVI	Single dose 1 g IVI if surgery > 4 h
	If allergic: Vancomycin	30 mg/kg slowly IVI over 120 min	Single dose
Cranio-cerebral wounds	Peni A + IB[b]	2 g slow IVI	2 g every 8 hours 48 h maximum
	If allergic: Vancomycin	30 mg/kg slowly IVI over 120 min	Single dose
Base of skull fracture with rhinorrhoea	No antibiotic prophylaxis		

a: Indications of vancomycin: allergy to beta-lactams; suspected or proven colonisation by methicillin-resistant *staphylococcus,* re-operation in a patient hospitalised in a unit with an ecology including methicilline-resistant *Staphylococcus aureus*, previous antibiotic therapy. The injection lasts 120 minutes and must end at the latest at the beginning of the intervention and the best 30 minutes before.
b: Aminopenicillin + beta-lactamase inhibitor.

Table II: Ophthalmology

SURGERY	PRODUCT	INITIAL DOSE	DOSAGE AND DURATION
Open eye surgery other than cataracts with risk factors	Levofloxazin oral dose	500 mg po	1 tab po 12 hours pre-operative + 1 tab po 2–4 hours pre-operative
Cataract[a]	Intracameral injection cefuroxime[a]	1 mg in 0.1 ml	At the end of surgery
Open eye trauma	Levofloxazin	500 mg	500 mg IV on day 1 + 500 mg orally on day 2
Lacrimal duct wounds	Peni A + IB[b]	2 g	Re-injection of 1 g if surgery > 2 hours
Puncture of anterior chamber Sub-retinal fluid puncture Closed globe surgery Intravitreal injections	No antimicrobial prophylaxis		

a: For cataract surgery with and without risk factors, a single injection into the anterior chamber of cefuroxime (1 mg) has been approved since 2014.
b: Aminopenicillin + beta-lactamase inhibitor.

Table III: Cardiac surgery

SURGERY	PRODUCT	INITIAL DOSE	RE-INJECTION DOSE AND DURATION
Cardiac surgery	Cefazolin	2 g IVI and 1 g in priming	1 g at the 4th hour intraoperatively
	Cefamandole or cefuroxime	1.5 g IV + 0.75 g priming	1 re-injection 0.75 g every 2 hours intraoperatively
	If allergic: Vancomycin[a]	30 mg/kg/120 min	Single dose
Pacemaker Endovascular procedures	See above heart surgery		Single dose
Pericardial drainage Coronary dilatation/stent ECMO	No antimicrobial prophylaxis		

a: Indications of vancomycin: allergy to beta-lactams; suspected or proven colonisation by methicillin-resistant *staphylococcus*, re-operation in a patient hospitalised in a unit with an ecology including methicilline-resistant *Staphylococcus aureus*, previous antibiotic therapy. The injection lasts 120 minutes and must end at the latest at the beginning of the intervention and the best 30 minutes before.

Table IV: Vascular surgery

SURGERY	PRODUCT	INITIAL DOSE	RE-INJECTION DOSE AND DURATION
Surgery of the aorta, arteries of the lower limbs, supra-aortic trunks Arterial endoprosthesis Carotid surgery with patch	Cefazolin	2 g slow IVI	Single dose (if time > 4 h, re-inject 1 g)
	Cefuroxime	1.5 g slow IVI	Single dose (if duration > 2 h, re-inject 0.75 g)
	If allergic: Vancomycin[a]	30 mg/kg/120 min	Single dose
Carotid surgery without a patch	No antimicrobial prophylaxis		
Expansion with or without stent	See above aortic surgery		Single dose
Limb amputation	Peni A + IB[b]	2 g IV slow	1 g/6 hours for a period of 48 hours
Vein surgery	No antimicrobial prophylaxis		

a: Indications of vancomycin: allergy to beta-lactams; suspected or proven colonisation by methicillin-resistant *staphylococcus*, re-operation in a patient hospitalised in a unit with an ecology including methicilline-resistant *Staphylococcus aureus*, previous antibiotic therapy. The injection lasts 120 minutes and must end at the latest at the beginning of the intervention and the best 30 minutes before.
b: Aminopenicillin + beta-lactamase inhibitor.

Table V: Orthopaedic surgery

SURGERY	PRODUCT	INITIAL DOSE	RE-INJECTION DOSE AND DURATION
Joint prosthesis (upper limb, lower limb)	Cefazolin	2 g slow IVI	1 g if duration > 4 h Limited to the operative period (24 hours max)
	Cefuroxime	1.5 g IV slow	0.75 g if duration > 2 h Limited to the operative period (24 hours max)
	If allergic: Clindamycin	900 mg IV slow	Limited to the operative period (24 hours max)
	If allergic: Vancomycin[a]	30 mg/kg/120 min	Single dose
Foreign material (resorbable or not, cement, bone graft) and whatever the technique (percutaneous, videoscopy) Joint surgery arthrotomy	Cefazolin	2 g slow IVI	1 g if duration > 4 h
	If allergic: Clindamycin	900 mg slow IVI	Single dose
	or Vancomycin[a]	30 mg/kg/120 min	Single dose
Arthroscopy without implant (with or without meniscectomy) extra-articular soft tissue surgery without implant	No antimicrobial prophylaxis		
Spine surgery with implantation of prosthetic material	Cefazolin	2 g slow IVI	Single dose (if duration > 4 h re-inject 1 g)
	If allergic: Vancomycin[a]	30 mg/kg/120 min	Single dose

a: Indications of vancomycin: allergy to beta-lactams, suspected or proven colonisation by methicillin-resistant *staphylococcus*, re-operation in a patient hospitalised in a unit with an ecology including methicilline-resistant *Staphylococcus aureus*, previous antibiotic therapy. The injection lasts 120 minutes and must end at the latest at the beginning of the intervention and the best 30 minutes before.

Table VI: Trauma

SURGERY	PRODUCT	INITIAL DOSE	RE-INJECTION DOSE AND DURATION
Closed fracture requiring isolated extrafocal osteosynthesis	No antimicrobial prophylaxis		
Closed fracture requiring intrafocal osteosynthesis Open fracture stage I Cauchoix Soft tissue wound non-contused, with or without lesion of artery, nerve, tendon Articular wound	Cefazolin	2 g slow IVI	1 g if duration > 4 h Limited to the operative period (24 hours max)
	Cefuroxime	1.5 g slow IVI	0.75 g if duration > 2 h Limited to the operative period (24 hours max)
	If allergic: Clindamycin *and* Gentamycin	900 mg slow IVI 5 mg/kg/day	600 mg if duration > 4 h
Open fracture stage II and III Cauchoix Large wound soft tissue contused with or without lesion of artery, nerve, tendon	Peni A + IB[a]	2g slow IVI	1 g if duration > 2 h 48 h maximum
	If allergic: Clindamycin *and* Gentamycin	900 mg slow IVI 5 mg/kg/day	600 mg if duration > 4 h 48 h maximum Max 48 hours

a: Aminopenicillin + beta-lactamase inhibitor.

Table VII: Thoracic surgery

SURGERY	PRODUCT	INITIAL DOSE	RE-INJECTION DOSE AND DURATION
Pulmonary resection (including video-assisted surgery)	Peni A + IB[a]	2 g slow IVI	Single dose (if duration > 2 h re-inject 1 g)
	or Cefuroxime	1.5 g slow IVI	Single dose (if duration > 2 h re-inject 0.75 g)
	or Cefazolin	2 g slow IVI	Single dose (if surgery > 4 hours, re-inject 1 g)
	If allergic: Clindamycin *and*	900 mg slow IVI	Single dose (if surgery > 4 hours, re-inject 600 mg
	Gentamycin	5 mg/kg/day	Single dose
Mediastinal surgery Surgery for pneumothorax Decortication (uninfected patient) Isolated parietal resection	Cefuroxime	1.5 g slow IVI	Single dose (if duration > 2 h re-inject 0.75 g)
	or Cefazolin	2 g slow IVI	Single dose (if surgery > 4 hours, reinject 1 g)
	If allergic: Clindamycin *and*	900 mg slow IVI	Single dose (if surgery > 4 hours, re-inject 600 mg)
	Gentamycin	5 mg/kg/day	Single dose
Mediastinotomy, videothoracoscopy, tracheostomy, thoracic drainage	No antimicrobial prophylaxis		

a: Aminopenicillin + inhibitor of beta-lactamases.

Table VIII: ENT

SURGERY	PRODUCT	INITIAL DOSE	RE-INJECTION DOSE AND DURATION
Rhinologic surgery with placement of a graft or re-operation	Cefazolin	2 g slow IVI	Single dose
	Peni A + IB[a]	2 g slow IVI	Single dose
Neck surgery with oropharyngeal opening. Surgery of the salivary glands with access through the oropharyngeal cavity	Peni A + IB[a]	2 g slow IVI	Re-injection of 1 g every 2 h during intraoperative then 1 g every 6 hours for 24 hours
	If allergic: Clindamycin *and*	900 mg slow IVI	Re-injection of 600 mg if duration > 4 h and then 600 mg/6 hours for 24 hours
	Gentamycin	5 mg/kg/day	Single dose
Alveolar surgery	Prevention of endocarditis (see endocarditis prophylaxis)		
Stapes surgery, middle ear surgery Salivary surgery without access through the oropharyngeal cavity Cervicotomy Lymphadenectomy Velopalatin surgery Tonsillectomy	No antimicrobial prophylaxis		

a: Aminopenicillin + inhibitor of beta-lactamases.

Table IX: Maxillo-facial surgery

SURGERY	PRODUCT	INITIAL DOSE	RE-INJECTION DOSE AND DURATION
Maxillofacial surgery with oropharyngeal opening. Surgery of the salivary glands with access through the oropharyngeal cavity	Peni A + IB[a]	2 g slow IVI	Re-injection of 1 g every 2 h during intraoperative then 1 g every 6 hours for 24 hours
	If allergic: Clindamycin *and*	900 mg slow IVI	Re-injection of 600 mg if duration > 4 h and then 600 mg/6 hours for 24 hours
	Gentamycin	5 mg/kg/day	Single dose
Alveolar surgery	Prevention of endocarditis (see endocarditis prophylaxis)		
Salivary surgery without access through the oropharyngeal cavity Cervicotomy; Lymphadenectomy Velopalatin surgery	No antimicrobial prophylaxis		
Tooth extraction in a non-septic environment	Prevention of endocarditis (see endocarditis prophylaxis)		

a: Aminopenicillin + inhibitor of beta-lactamases.

Table X: Digestive surgery

SURGERY	PRODUCT	INITIAL DOSE	RE-INJECTION DOSE AND DURATION
Oesophageal surgery (without coloplasty) Gastroduodenal surgery (including endoscopic gastrostomy and duodenopancreatectomy) Pancreatic surgery Liver surgery	Cefazolin	2 g slow IVI	Single dose (if duration > 4 h re-inject 1 g)
	Cefuroxime	1.5 g slow IVI	Single dose (if duration > 2 h re-inject 0.75 g)
	If allergic: Clindamycin *and* Gentamycin	900 mg slow IVI 5 mg/kg/day	Single dose (if duration > for 4 h, inject 600 mg) Single dose
Biliary tract surgery (the biliary prosthesis patients are excluded from recommendations)	Cefazolin	2 g slow IVI	Single dose (if duration > 4 h re-inject 1 g)
	Cefuroxime	1.5 g slow IVI	Single dose (if duration > 2 h re-inject 0.75 g)
	If allergic: Clindamycin *and* Gentamycin	900 mg slow IVI 5 mg/kg/day	Single dose (if duration > for 4 h, inject 600 mg) Single dose
Gallbladder surgery laparoscopically without risk factors[a]	No antimicrobial prophylaxis		
Colonic and intestinal surgery	Cefoxitin *and* Metronidazole	2 g slow IVI Slow 1 g infusion	Single dose (if duration > 2 h re-inject 1 g) Single dose
	If allergic: Imidazole *and* Gentamycin	1 g infusion 5 mg/kg/day	Single dose Single dose
Anal surgery	Imidazole	1 g infusion	Single dose
Hernia without prosthetic plate	No antimicrobial prophylaxis		
Hernia with establishment of a prosthetic plate	Cefazolin	2 g slow IVI	Single dose (if duration > 4 h re-inject 1 g)
	Cefuroxime	1.5 g slow IVI	Single dose (if duration > 2 h re-inject 0.75 g)
	If allergic: Clindamycin *and* Gentamycin	900mg slow IVI 5mg/kg/day	Single dose (if duration > for 4 h, inject 600 mg) Single dose
Eventration	Cefazolin	2 g slow IVI	Single dose (if duration > 4 h re-inject 1 g)
	Cefuroxime	1.5 g slow IVI	Single dose (if duration > 2 h re-inject 0.75 g)
	If allergic: Clindamycin *and* Gentamycin	900 mg slow IVI 5 mg/kg/day	Single dose (if duration > for 4 h, inject 600 mg) Single dose
Abdominal wounds	See colorectal surgery		
Prolaps (any surgical approach)	Peni A + IB[b]	2 g slow IVI	Single dose 1 g if duration > 2 h
	If allergic: Gentamycin *and* Metronidazole	5 mg/kg/day Slow 1 g infusion	Single dose Single dose

a: Laparoscopic cholecystectomy without risk factors: absence of recent cholecystitis, no conversion to laparotomy (on the event of conversion to ABP), no pregnancy, no immune suppression, no exploration of bile duct intraoperatively. If risk factors, refer to the "biliary tract surgery".
b: Aminopenicillin + beta-lactamase inhibitor.

Table XI: Urology

SURGERY	PRODUCT	INITIAL DOSE	RE-INJECTION DOSE AND DURATION
Prostate surgery			
Endoscopic resection of the prostate, cervical-prostatic incision prostatectomy	Cefazolin	2 g slow IVI	Single dose (if duration > 4 h re-inject 1 g)
	Cefuroxime	1.5 mg slow IVI	Single dose (if duration > 2 h, re-inject 0.75 g)
	If allergic:		
	Gentamycin	5 mg/kg	Single dose
Radical prostatectomy	No antimicrobial prophylaxis		
Prostate biopsy	Ofloxazin	400 mg 1 hour before the biopsy	Single dose
	If allergic:		
	Ceftriaxone	1 g	Single dose
Kidney surgery, adrenal and urinary tract			
Endoscopic treatment of the renal and ureteral ithiases, ureteroscopy, percutaneous nephrolithotomy, nephrostomy, JJ probe mounted or ureteral	Cefazolin	2 g slow IVI	Single dose (if duration > 4 h re-inject 1 g)
	Cefuroxime	1.5 mg slow IVI	Single dose (if duration > 2 h, re-inject 0.75 g)
	If allergic:		
	Gentamycin	5 mg/kg	Single dose
Transurethral resection of the prostate	Cefazolin	2 g slow IVI	Single dose (if duration > 4 h re-inject 1 g)
	Cefuroxime	1.5 mg slow IVI	Single dose (if duration > 2 h, re-inject 0.75 g)
	If allergic:		
	Gentamycin	5 mg/kg	Single dose
Nephrectomy and other upper tract surgery Extracorporeal lithotripsy Adrenalectomy	No antimicrobial prophylaxis		
Bladder surgery			
Transurethral resection of the prostate	Cefazolin	2 g slow IVI	Single dose (if duration > 4 h re-inject 1 g)
	Cefuroxime	1.5 mg slow IVI	Single dose (if duration > 2 h, re-inject 0.75 g)
	If allergic:		
	Gentamycin	5 mg/kg	Single dose
Cystectomy (Bricker, bladder replacement)	PENI A + IB[a]	2 g slow IVI	Single dose (if duration > 2 h re-inject 1 g)
	If allergic:		
	Gentamycin	5 mg/kg	Single dose
	and		
	Metronidazole	Slow 1 g infusion	Single dose
Surgery of the urethra			
Urethroplasty, urethrotomy	Cefazolin	2 g slow IVI	Single dose (if duration > 4 h re-inject 1 g)
	Cefuroxime	1.5 mg slow IVI	Single dose (if duration > 2h, re-inject 0.75 g)
	If allergic:		
	Gentamycin	5 mg/kg	Single dose
Artificial sphincter	Cefoxitin *or* Amoxicillin + Clavulanic acid	2 g slow IVI	Single dose
	If allergic:		
	Gentamycin	5 mg/kg	Single dose
	and		
	Metronidazole	1 g infusion	
Urethral support (TOT, TVT)	PENI A + IB[a]	2 g slow IVI	Single dose
	If allergic:		
	Gentamycin	5 mg/kg	
	and		
	Metronidazole	1 g infusion	
Male genital system			
Scrotal surgery or rod (not replacement)	No antimicrobial prophylaxis		
Penile prosthesis or testicular	Cefazolin	2 g slow IVI	Single dose (if surgery > 2 hours, re-inject 1 g)
Female genital system			
Treatment of prolapse (any surgical approach)	PENI A + IB[a]	2 g slow IVI	Single dose (if surgery > 2 h, re-inject 1 g)
	If allergic:		
	Metronidazole	1 g infusion	Single dose
	and		
	Gentamycin	5 mg/kg/day	Single dose
Diagnostic investigations Bladder fibroscopy Urodynamic evaluation Ureteroscopic diagnostic	No antimicrobial prophylaxis		

a: Aminopenicillin + beta-lactamase inhibitor.

Table XII: Gynaecology obstetrics

SURGERY	PRODUCT	INITIAL DOSE	RE-INJECTION DOSE AND DURATION
Hysterectomy (high or low road)	Cefazolin	2 g slow IVI	Single dose (if surgery > 4 hours, re-inject 1 g)
Coelio surgery (Laparoscopic surgery)	Cefuroxime	1.5 mg slow IVI	Single dose (if surgery > 2 hours, re-inject 0.75 g)
	If allergic:		
	Clindamycin	900 mg slow IVI	Single dose
	and		
	Gentamycin	5 mg/kg	Single dose
Laparoscopy diagnostic or exploratory without vaginal incision or digestive	No antimicrobial prophylaxis		
Hysteroscopy hysterosalpingography	No antimicrobial prophylaxis		
Endometrial biopsy	No antimicrobial prophylaxis		
In vitro fertilisation	No antimicrobial prophylaxis		
Insertion of intra-uterine device	No antimicrobial prophylaxis		
Abortion	No antimicrobial prophylaxis		
Caesarean section	Cefazolin	2 g slow IVI	Single dose
	Cefuroxine	1.5 g slow IVI	Single dose
	If allergic:		
	Clindamycin	900 mg slow IVI	Single dose
Mastectomy Reconstruction and/or mammoplasty	Cefazolin	2 g slow IVI	Single dose (1 g if duration > 4 hours)
	Cefuroxine	1.5 g slow IVI	Single dose (0.75 g if surgery > 2 hours)
	If allergic:		
	Clindamycin	900 mg slow IVI	600 mg if surgery > 4 hours
	and		
	Gentamycin	5 mg/kg	Single dose
Simple breast lumpectomy	No antimicrobial prophylaxis		
Prolapse (all surgical approaches; only in case of implementation of prosthetic material: promontofixation, implant placement or strip)	Peni A + IB[a]	2 g slow IVI	Single dose (1 g if surgery > 4 hours)
	If allergic:		
	Gentamycin	5 mg/kg	Single dose
	and		
	Metronidazole	1 g infusion	Single dose

a: Aminopenicillin + beta-lactamase inhibitor.

Table XIII: Interventional surgery

SURGERY	PRODUCT	INITIAL DOSE	RE-INJECTION DOSE AND DURATION
Embolisation of uterine fibroids	No antimicrobial prophylaxis		
Transjugular intrahepatic portosystemic shunt	No antimicrobial prophylaxis		
Nephrostomy	No antimicrobial prophylaxis		
Endoscopic gastrostomy, sclerosis of oesophageal varicose veins	Peni A +1B[a]	2 g slow IVI	Single dose
	If allergic:		
	Clindamycin	900 mg slow IVI	Single dose
	and		
	Gentamycin	5 mg/kg	Single dose
Puncture under endoscopic ultrasonography (except trans-colorectal puncture)	No antimicrobial prophylaxis		
Endoscopic dilatation, digestive prosthesis, laser, argon, plasma coagulation	No antimicrobial prophylaxis		
Chemo-embolisation	No antimicrobial prophylaxis		
Radio frequency	No antimicrobial prophylaxis		
Catheter or implantable chamber	No antimicrobial prophylaxis		
Angiography/angioplasty	No antimicrobial prophylaxis		
Stent (excluding intra-coronary)	Cefazolin	2 g slow IVI	Single dose (if duration > 4 hours, re-inject 1 g)
	Cefuroxime	1.5 g slow IVI	Single dose (if duration > 2 hours; re-inject 0.75 g)
	If allergic:		
	Vancomycin[b]	30 mg/kg/120 min	Single dose

a: Aminopenicillin + beta-lactamase inhibitor.
b: Indications of vancomycin: allergy to beta-lactams; suspected or proven colonisation by methicillin-resistant *staphylococcus*, re-operation in a patient hospitalised in a unit with an ecology including methicillin-resistant *Staphylococcus aureus*, previous antibiotic therapy. The injection lasts 120 minutes and must end at the latest at the beginning of the intervention and the best 30 minutes before.

Table XIV: Bariatric surgery

SURGERY	PRODUCT	INITIAL DOSE	RE-INJECTION DOSE AND DURATION
Gastric band	Cefazolin	4 g as an infusion over 30 min	Single dose (if duration > 4 hours, inject 2 g)
	Cefuroxime	3 g as an infusion over 30 min	Single dose (if duration > 2 hours, inject 1.5 g)
	If allergic:		
	Vancomycin[a]	30 mg/kg/120 min	Single dose
Performing a gastric bypass or "sleeve" gastrectomy	Cefoxitin	4 g (30 min infusion)	Single dose (if duration > 2 hours, reinject 2 g)
	If allergic:		
	Clindamycin	2 100 mg slow IVI	Single dose
	and		
	Gentamycin	5 mg/kg	Single dose
Abdominoplasty (dermolipectomy)	Cefazolin	4 g (30 min infusion)[b]	Single dose (if duration > 4 hours, inject 2g)
	Cefuroxime	3 g (30 min infusion)[b]	Single dose (if duration >2 hours, inject 1.5 g)
	If allergic:		
	Clindamycin	2 100 mg slow IVI	Single dose
	and		
	Gentamycin	5 mg/kg (dose based on actual weight)	Single dose

a: Indications of vancomycin: allergy to beta-lactams, suspected or proven colonisation by methicillin-resistant *staphylococcus*, re-operation in a patient hospitalised in a unit with an ecology including methicillin-resistant *Staphylococcus aureus*, previous antibiotic therapy. The injection is done at 1 000 mg/hour maximum and must end at the latest at the beginning of the intervention and at best 30 minutes before. Maximum dose is 4 g.
b: Dose calculated on the actual weight.

Table XV: Plastic and reconstructive surgery

SURGERY	PRODUCT	INITIAL DOSE	RE-INJECTION DOSE AND DURATION
Plastic and reconstructive surgery: class 1 Altemeier	No antimicrobial prophylaxis in the absence of implant		
	Cefazolin	2 g slow IVI	Single dose (if duration > 4 hours, re-inject 1 g)
	If allergic:		
	Clindamycin	900 mg slow IVI	Single dose (if duration > 4 hour, re-inject 600 mg)
Plastic and reconstructive surgery: class 2 Altemeier	Cefazolin	2 g slow IVI	Single dose (if duration > 2 hours, re-inject 1 g)
	If allergic:		
	Clindamycin	900 mg slow IVI	Single dose (if duration > 4 hour, re-inject 600 mg)

Table XVI: Prophylaxis of endocarditis

Antibiotics 30–60min prior to the procedure			
SURGERY	**PRODUCT**	**ADULT**	**CHILDREN**
No allergy to beta-lactams	Amoxicillin or ampicillin	2 g oral or IVI	50 mg/kg orally or IVI
Allergy to beta-lactams	Clindamycin	600 mg orally or IVI	20 mg/kg orally or IVI

Chapter 10: Environmental considerations in infection control and prevention

Please refer to the relevant IUSS guidelines that can be found at www.iussonline.co.za as well as the SASA website

Operating theatre ventilation[263-265]

- Every operating theatre complex requires an effective central humidity ventilation and air-conditioning system (HVAC).
- Maintain at least 15–20 air changes per hour of which at least three such air changes should be with fresh air.
- Maintain positive pressure within the operating theatre compared to the corridors and adjacent areas. This is to facilitate the movement of air from the operating theatre along a pressure gradient towards adjacent areas. Air thus moves away from the sterile operating site, thus reducing the risk of contaminated air getting in contact with the operating field and sterile instruments.
- Ideally, air should come from the ceiling and move towards at least two air exhaust vents located close to the floor.
- Make sure that air exhaust vents are not obstructed or covered.
- All air (re-circulating and fresh air) should pass through filters before entering the operating theatre.
- Keep the doors to the operating theatres closed at all times.
- The air-conditioner system should operate continuously independent of whether there is a case on the table or not.

Rationale[263]

The quality of air in theatre is important both for the comfort of the personnel but more importantly to prevent contamination of the surgical wound and/or transmission of pathogens. Air in theatre can contain skin squames from both patient and healthcare workers, respiratory droplets and aerosols that may contain microorganisms, dust, lint and anaesthetic gasses. Airflow should be directed in such a fashion that it will cause the cleanest air to be closest to the surgical wound and dirty air to move away from the wound. Clean air coming from a ceiling pendant and moving towards air exhaust vents close to the floor can aid in this regard. The velocity of the air flow should be low to avoid drafts and swirls that can lead to the re-circulation of microbes. It should, however, be high enough to allow adequate air changes per hour. The air-conditioner system should operate continuously, even if there is no case being done. This is to prevent the theatre from being contaminated during down-time.

Forced-air warmers

- Consider using conductive fabric warmers over forced-air warmers for preventing intraoperative hypothermia, especially in high-risk surgery such as joint replacements.[265,266]
- Where conductive fabric warmers are not available, forced-air warmers should be used as the risk of hypothermia outweighs the possible risk of contamination of the surgical wound.

Rationale

The use of forced-air warming blankets is common practice to prevent intraoperative hypothermia. McGovern et al.[266] had looked at air currents in a simulated scenario in theatres with ultra-clean ventilation. They used neutral bubbles to allow visualisation of air currents. The use of a forced-air warming blanket was associated with contamination of the surgical field by mobilising particles from under the drapes. The machine that generates a forced-air current is usually placed on the floor. It creates a warm area that results in convection currents. It was shown that these convection currents cause an upward air current with mobilisation of particles from the floor. These particles contaminated the surgical site. A review of the literature by Kellam et al.[267] found that current evidence is inconclusive with regards to the actual risk of surgical site infections. It is also unclear whether the type of ventilation makes a difference with Sessler et al.[268] finding that air currents are not affected by a forced-air warming device when used in a laminar flow theatre. More high quality studies are needed before a definitive recommendation can be made.

Operating theatre temperature and humidity[264,269]

- Keep ambient theatre temperature between 18–24 ºC.
- The ambient temperature should be uniform throughout the space.
- Keep humidity levels between 30–60%.

Rationale[263]

The international standard for ambient theatre temperature is between 20–24 ºC. The South African IUSS standards recommend temperatures to be between 18–24 °C. There may be certain cases where temperatures outside of this range may be indicated. Higher ambient temperatures are needed in theatres where burns cases or paediatric cases are being done. It should be noted that this is a trade-off with an increased risk of infection. Cases that may require a lower temperature include cardiac theatres and orthopaedic theatres, especially theatres where arthroplasty cases are done.

Humidity should be maintained within the recommended range. Humidity has a direct influence on thermal comfort. In addition, it influences the quality of air with regards to the microbial load.

Low humidity environments increase the risk that microorganisms especially Gram-positive cocci and fungi that can persist with dust particles or on surfaces.[270] Enterococci can persist on dry areas for between seven days to four months.[271] High humidity can be uncomfortable to theatre staff and it also promotes the growth of microorganisms, especially Gram-negative bacilli and fungi. Microorganisms trapped in the water molecules can become airborne and carried over larger areas.[263]

Environmental cleaning of the operating theatre[70,269,272]

Definitions:

Cleaning	• The physical removal of foreign material, e.g. dust, soil, and organic material such as: blood, secretions, excretions and microorganisms. Cleaning physically removes rather than kills microorganisms. • It is accomplished with water, detergents and mechanical action.
Contact time/ Dwell time	• The defined time for which surfaces are exposed to a chemical or thermal disinfection process to achieve the appropriate level of disinfection. • Inadequate contact time may lead to incomplete disinfection.
Disinfectant	• Product used on inanimate objects to reduce the quantity of microorganisms to an acceptable level. • Hospital-grade disinfectants require a drug identification number (DIN). • High-level disinfectants should not be used to clean environmental or inanimate objects.
Disinfection	• The inactivation of disease-producing microorganism with the exception of bacterial spores. • Hospital-grade disinfectants are used on inanimate objects. • Medical equipment must be cleaned properly before effective disinfection can take place.
Preliminary cleaning	• Damp dust horizontal surfaces prior to first case. Do not clean with dry materials as that causes dust to become airborne. • Use a clean, lint-free cloth moistened with low-level disinfectant. • Avoid spraying or misting methods. • Start at higher surfaces and work down in a clockwise manner. • Damp dust equipment before it is brought into or out of the operating theatre. • Inspect operating theatre lights for cleanliness before the first case of the day. • Floors should always be considered contaminated even after proper cleaning.
Intraoperative cleaning	• The responsibility for verifying disinfection of a contaminated surface rests with the perioperative team member who is first aware of the contamination. • All contaminated (blood, body fluids, or other potentially infectious material) items or surfaces occurring intraoperatively are to be promptly cleaned/disinfected as required using facility-approved disinfectant. • Equipment leaving the operating theatre is cleaned and disinfected with hospital approved disinfectant. • Chemical spills occurring intraoperatively are to be managed as per site/regional policy/procedure.
Between procedures	• Each operating theatre must be cleaned and disinfected immediately after each case. • Do not start the process before the patient has left the area. • Prior to cleaning, remove all trash, linen, and recycling from the room, including soiled anaesthesia equipment and supplies. • All surfaces that have been in direct or indirect contact with the patient or body fluids are considered to be contaminated and therefore are to be cleaned/disinfected with a hospital-approved disinfectant. • Contaminated linen should be handled as little as possible.
Terminal cleaning	• Cleaning staff should adhere to standard precautions with regards to PPE. • Staff preforming cleaning may be required to wear additional PPE during terminal cleaning after procedures with additional precautions such as the highly infectious patient. • Operating theatres are to be terminally cleaned at minimum once every 24 hours during a regular workweek regardless of whether the theatre has been used. • All floors should be cleaned using a wet vacuum or single-use mop and a disinfectant (follow dwell time indicated on manufacturer's instructions). • Floor cleaning should progress from cleanest area to dirtiest, from perimeter of the room to the centre. • Care must be taken to ensure the floor under the theatre bed and trolleys are also cleaned.

Cleaning of surfaces and instruments with detergents is needed before disinfection and decontamination can take place. The presence of organic matter, salts and obvious contamination can compromise the efficacy of the terminal reprocessing procedures.

Use EPA-approved disinfectants such as a quaternary ammonium compound to clean all areas that could have been contaminated during the procedure. Avoid methods that can lead to mist, aerosols or dispersion of dust.

Some centres use hydrogen peroxide automated fogging machines. The operating theatre is sealed and locked while the machine is in use. The theatre can be entered once the hydrogen peroxide level has decreased to less than 1 ppm. Decreases in microbial load are in the region of 4 logs.[273,274] Ultraviolet light is also being used and seems to have a log reduction of 1–2 logs.

Surgical attire[269,275-278]

Scrubs

- Scrubs should be made from material that is tightly woven, low-linting, stain-resistant and non-flammable.
- It should not be 100% fleece but rather a mixture of cotton with 10–20% polyester.
- Change scrubs that are visibly soiled or contaminated with potentially infectious material.
- Reusable scrubs should be washed at a healthcare-accredited laundry.
- For procedures at high risk of contamination from blood or bodily fluids, a waterproof apron should be worn under the surgical gown.
- Surgical scrubs should not be worn outside the operating theatre complex due to the risk of spreading microorganisms.

- Home laundering of scrubs is not recommended as it does not meet specified criteria to reduce microbial load.
- Maintaining good personal hygiene is as important as wearing appropriate theatre attire.

Other theatre attire

- Wear a surgical mask when packs are open and for the duration of the procedure.
- The surgical mask must cover all facial hair, the mouth and the nose.
- Wear a theatre cap that covers all hair on the head and sideburns.
- Shoe covers do not protect against SSIs.
- Shoe covers can be replaced with dedicated theatre shoes that are easily washable and washed at the end of the day.
- Outside shoes should not be worn inside the operating theatre.
- PPE use should be based on the most likely mode of transmission of organisms.

Rationale

Members of the surgical team entering the operating theatre when an operation is about to begin or already underway should wear a mask and headgear that fully covers hair, sideburns, and neckline. Experimental studies using tracer particles have shown that bacteria can be shed from hair, exposed skin, and mucous membranes of both operating theatre personnel and the patient's skin. This is why we use barriers (masks, gowns, hood, and drapes) in the operating theatre. Besides sterile gloves and impervious surgical gowns, no clinical studies have proved that the use of these barriers has led to a decrease in SSI rates.[279] They are nonetheless recommended not only for the purpose of reducing the shedding of microorganisms in the operating theatre but also as part of standard precautions. Barriers are most important when the procedure implies the insertion of an implant/prosthesis.

The type of surgical headgear (bouffant or skull caps) has been called into question. A 2017 study showed that disposable bouffant hats had greater permeability and microbial shed and increased passive microbial shed compared to disposable skull caps and cloth caps.[280] Multiple studies have shown the hair of personnel to be contaminated with microorganisms, especially *Staphylococcus Aureus* and *Coagulase-negative staphylococcus*.[281-283]

Floors are rapidly re-contaminated after being washed. Shoes and shoe covers subsequently become contaminated and these microorganisms can then be transferred from patient to bed sheets or to hands when shoe covers are removed.[284,285] Shoe covers can be replaced by ordinary shoes dedicated exclusively to the operating theatre because no significant difference was found in floor contamination whether personnel wear shoe covers or dedicated theatre shoes. These shoes must be easy to wash. Shoes worn outside theatre can be visibly contaminated and should be changed for dedicated theatre shoes. The practice of wearing plastic/paper shoe covers for the purpose of decreasing SSIs should be abandoned.

Personal protective equipment

- Different types of protective clothing are needed in different parts of the perioperative environment.
- PPE are different barriers that are used in a combination to protect skin, mucosa, breathing ways, and clothes from coming into contact with blood-borne pathogens, other potentially infected materials, and chemicals. PPE includes gown or apron, gloves, mask, goggles, and visor.
- Standard precautions should always be applied, and additional precautions should be based on mode of transmission.
- They are also used to protect the instruments from getting contaminated by bacteria and other particles from the human body. Due to the shedding of bacteria from the human body, the use of protective clothing, as well as personal hygiene, are of great importance when it comes to the control of cross-infection risks.

Chapter 11: Infection control precautions for the infectious patient

Non-emergency cases should be postponed until the patient is deemed to no longer be infectious. Units should develop their own facility-based protocols. Precautions should be based on the mode of transmission of the specific microorganism.

Modes of transmission[36]

Direct contact

- Physical contact with patient and body fluid, soil or vegetation
- Droplet spread:
  - Large particles > 5 µm.
  - Short distance spread usually within one meter but can be further as has been demonstrated with smallpox and SARS viruses.
  - Airway instrumentation such as intubation, suctioning and extubation.
  - Sneezing, coughing, singing.

Indirect contact

- Airborne/aerosol spread:[286]
  - Particles are less than 5 µm.
  - Can remain suspended in air for prolonged periods.
  - Can spread over large areas and possibly further than physical barriers such as rooms or operating theatres.

- Can be deposited on environmental surfaces. Depending on the type of surface, it can remain viable for a couple of hours or up to a couple of days.
- Small size means the particles can travel further down the respiratory tract into alveoli.
- Aerosol-generating procedures (AGP):[287]
  - Airway instrumentation:
    - Intubation, extubation
    - Manual mask ventilation prior to intubation
    - Tracheostomy
    - Suctioning of airways (unless a closed system is used)
    - Placement of supraglottic airway device
    - Bronchoscopy
  - Chest physiotherapy
  - Nebulisation
  - Non-invasive ventilation including high nasal flow oxygen
  - Cardio-pulmonary resuscitation
  - Surgeries where high speed devices are used
  - Plume from cauterisation
  - Sneezing, coughing, vocalisation
  - Autopsy
- Vehicle spread:
  - Contaminated hands.
  - Inanimate objects such as laryngoscopes, pens, cell phones.
- Vector spread:
  - E.g. flies and mosquitoes.

Type of precautions

Standard precautions[36,288,289]

- Hand hygiene.
- Use of PPE.
- Respiratory etiquette.
- Environmental cleaning and disinfection.
- Proper handling of patient care equipment and waste management.
- Proper handling of needles and sharps.
- Healthcare workers should have documented immunity to hepatitis B virus.

Contact precautions[36,288,289]

- Standard precautions as above.
- Adhere to hand hygiene standards.
- Isolation cubicle until no longer infectious.
- Use PPE including a gown.
- Remove PPE before leaving the immediate environment of the patient.
- Take care not to self-contaminate when removing PPE.
- Maintain contact precautions throughout the entire perioperative period.
- Appropriate environmental decontamination of the operating theatre at the end of the case.

Droplet precautions[36,288,289]

- Standard precautions as above.
- Adhere to hand hygiene standards.
- Ideally, to be isolated. If isolation not available, keep patient at least one meter from any other patients.
- Educate patients on respiratory hygiene, e.g. coughing and sneezing etiquette.
- Patient to wear a standard face mask when outside the isolation area.
- Healthcare workers must use standard PPE.
- Use N95 masks/respirators during AGP such as airway instrumentation.
- Maintain these precautions throughout the entire perioperative period.

Airborne precautions[36,288,289]

- Standard precautions as above.
- Adhere to hand hygiene standards.
- Patient should be in an airborne isolation room.
- Patient should remain in the isolation room except for medical/surgical procedures that require the patient to leave the room.
- Any non-emergency case should be postponed until the patient no longer needs respiratory isolation.
- Patient to wear a standard face mask when outside the isolation area.

Healthcare workers donning of PPE[290,291]

- There are various different guidelines that differ in the sequence of events. The most important thing to consider is to minimise the risk of contamination, especially during the doffing procedure. It is advisable to have a "buddy system" where a colleague uses a checklist to guide the sequence of events. In addition, it is valuable to choose one method and use that routinely to become familiar with such a method.
  - Remove jewellery and any other personal items.
  - Empty pockets.
  - Tie hair back and cover ALL hair.
  - Perform hand hygiene and don disposable apron.
  - Perform hand hygiene and change shoes or apply overshoes.
  - Perform hand hygiene and don first pair of gloves.
  - Perform hand hygiene and don an impermeable disposable gown or coverall.
    - Ensure that gown covers area from the neck to knees, arms to the end of wrists and wrapped around the back.
    - Tie behind with a simple knot that can be easily untied.
  - Perform hand hygiene and don respiratory protection
    - Use N95 or higher mask/respirator.
    - Secure ties at middle of head and neck.

- Fit flexible nose piece over the bridge of the nose.
- Fit snuggly to face and chin.
- Perform fit test: Mask collapses inward on inspiration and expands without a leak on exhalation.

◦ Hand hygiene and don goggles.
◦ Hand hygiene and don head cover or balaclava. Ensure that the sides of the goggles, the ears and nape of the neck are covered. Ideally, the neck should be covered as well.
◦ Hand hygiene and don second pair of gloves. Extend to cover wrist. No skin should be exposed.
◦ Hand hygiene and don face shield.

Healthcare workers doffing of PPE[290,291]

- Careful doffing is extremely important to prevent contamination.
  ◦ Use the method that you are familiar with.
  ◦ Perform hand hygiene on outer pair of gloves.
  ◦ Remove gown (Remember that the front of the gown and the arms are the most contaminated areas). Undo ties and carefully pull the gown down from neck and shoulders. Turn gown inside out.
  ◦ Outside gloves are removed with the gown as it reaches the wrist and hands.
  ◦ Carefully roll the gown with inside facing outwards into a bundle and discard.
  ◦ Perform hand hygiene on inner gloves using alcohol sanitiser.
  ◦ Remove face shield (remember that front is contaminated).
  ◦ Take off head covering – grasp from behind and carefully lift from the back of the head and into waste packet.
  ◦ Perform hand hygiene on inner gloves with alcohol sanitiser
  ◦ Carefully remove goggles.
  ◦ Hand hygiene.
  ◦ Remove overshoes.
  ◦ Hand hygiene.
  ◦ Exit doffing area and then remove N95 mask/respirator.
  ◦ Hand hygiene.
  ◦ Remove inner pair of gloves followed by hand hygiene.
- Maintain these precautions throughout the entire perioperative period.

Precautions for common pathogens

Pathogen	Level of precautions
HIV	Standard, post-exposure prophylaxis
Hepatitis (viral)	Standard precautions
Human papillomavirus (HPV)	Standard, droplet and airborne if plume from cauterisation of airway papillomas or genital warts are present
Measles	Standard and airborne
Mycobacterium tuberculosis	Standard and airborne precautions
Multidrug-resistant organisms Meticillin-resistant *Staphylococcus Aureus* (MRSA) Vancomycin-resistant enterococci (VRE) Extended-spectrum beta-lactamase-producing organisms (ESBL)	Standard and contact precautions
Neisseria	Standard and droplet precautions
Clostridium difficile	Standard and contact precautions
Haemophilus influenza	Standard and droplet precautions
Seasonal influenza	Standard and droplet precautions
Coronaviruses	Standard, droplet and airborne precautions

Rationale

Modes of transmission[36]

Microorganisms have different modes in which they are transmitted. These modes largely dictate what type of precautions should be adhered to in order to minimise the risk of transmission.

Direct contact occurs when there is physical contact with the organism, such as when there is contact with blood, body fluids or a contaminated environment. Droplet spread is a type of direct contact as the expelled droplets travel over a distance and make direct contact with the surrounding surfaces. Sneezing, coughing and talking are the most common ways in which droplets are generated. Procedures such as intubation, extubation, airway suctioning, cough induction by physiotherapist, and cardiopulmonary resuscitation can also produce large amounts of droplets and aerosols. Droplets are typically more than 5 µm and travel over short distances. This distance is generally considered to be about one meter but can be more than two meters. A study in Hong Kong has demonstrated the spread of SARS viruses to medical students that were further than one meter away from a single infected patient.[292] Factors that may contribute to droplets travelling further include the velocity of the expulsion of droplets (i.e. sneezing versus talking), the density of the respiratory secretions, ambient temperature and humidity as well as the ability of the microorganism to retain infectivity for longer periods.[36]

Indirect contact occurs when small particles become suspended in air currents. Droplet nuclei also become airborne pathogens. The size of these particles is generally smaller than 5 µm. They remain suspended for an undefined period of time. As such, they have the ability to spread over large areas and even past physical barriers such as rooms and operating theatres. Aerosoles can also be deposited on environmental surfaces and instruments such as laryngoscopes, pens, cell phones, etc. When such contaminated surfaces are touched, the microorganisms can then be transferred to hands that act as vehicles for the transmission of the organism. Vectors such as mosquitoes and flies are also examples of spread via indirect contact.

Precautions[36,289]

Standard precautions are applicable to any patient and not just the infectious patient. It involves following of all the guidelines set out in this document and specifically hand hygiene. In addition, healthcare workers should protect themselves where possible such as in the case with the hepatitis B vaccine.

Contact precautions include standard precautions with strict adherence to hand hygiene and donning and doffing of PPE. Care should be taken to avoid self-contamination when removing PPE. Contact precautions should remain in place for the entire time that the patient is occupying the operating theatre. PPE should be removed before leaving the immediate environment of the patient. Appropriate environmental decontamination should be performed after the patient has left the operating theatre.

The patient should further be nursed in an isolation cubicle until no longer deemed to be infectious. Whilst there is a major focus on droplet and airborne precautions, it is important to adhere to contact precautions, especially in those patients who may potentially harbour multidrug-resistant microorganisms. A recent study by Arriero et al.[293] showed the compliance rate of healthcare workers with empirical contact precautions to be 39.5% and 43.1% in patients known to be colonised. Compliance was better before patient contact than after, except for hand hygiene that showed better compliance after patient contact.

Droplet precautions include standard precautions with strict adherence to hand hygiene. PPE should be used when airway procedures are performed and should include a surgical mask and eye protection. MacIntyre et al.[294] looked at the efficacy of medical masks to reduce the risk of infection with influenza virus in healthcare workers. The continuous use of N95+ masks/respirators was associated with lower infection rates indicating that droplets can become aerosols even over short distances and that pathogens that are presumed to have droplet transmission only can also have airborne transmission. This sentiment is shared by Shiu et al.[288] This is especially important for AGPs. Anyone closer than one meter to the source patient should wear PPE, including surgical facemask. Proper airborne precautions should be in place for AGPS. Precautions should remain in place throughout the entire procedure. Organisms transmitted through droplets usually do not remain infective over long distances. Isolation is thus not crucial, but the patient should be more than one meter from other patients. Patients should also be educated on proper respiratory hygiene such as coughing and sneezing etiquette.

Airborne precautions are important as aerosols containing microorganisms can remain suspended in air for long periods and can travel over long distances. When these particles are inhaled, they colonise the naso- and oropharynx. Due to their small size, they also travel deep into the respiratory tree leading to lower respiratory tract infection and more severe disease. These patients should remain in an airborne isolation room until there is proof that they are no longer infectious. Non-emergency surgery should be postponed until the patient is no longer infectious. For emergency surgical procedures the patient should wear a surgical face mask before leaving the isolation cubicle. They should bypass the reception area and go straight to the operating theatre. All personnel, including porters, should wear fit tested N95 masks or respirators with more than 95% filtration in addition to standard PPE. These precautions should remain in place for the entire time where there is patient contact. All non-essential personnel should leave the operating theatre and non-essential trolleys and equipment should be removed. The patient should be recovered in theatre and returned straight to the isolation cubicle. The operating theatre should remain vacant after the source patient has left theatre until a 99.9% turnover of air has occurred. Air changes per hour differ between institutions and vary between 2–400 changes per hour. At two air changes per minute, 207 minutes will be required to ensure 99.9% removal of airborne particles. At the standard of 15 air changes per hour, a total of 28 minutes would result in 99.9% removal.[295,296] Cleaning of the operating theatre should only commence *after* this mandatory period has elapsed. Cleaners should also don proper PPE and should also be included in training on appropriate donning and doffing procedures.

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