



**SOUTH AFRICAN
SOCIETY OF
ANAESTHESIOLOGISTS
(SASA)**

SASA Guidelines for Infection Control in Anaesthesia in South Africa 2014

SASA Working Group for Infection Control Guidelines
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Guidelines for infection control 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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1. Introduction

“The very first requirement in a hospital is that it should do the sick no harm.” - Florence Nightingale

Notes on nursing: what it is, and what it is not

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 are 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 recent 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 that of many other countries. In addition, different regions face unique challenges within 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.

References

1. Smith CM. Origin and uses of *primum non nocere*: above all, do no harm! *J Clin Pharmacol.* 2005;45(4):371-377.
2. Brink A, Feldman C, Duse A, et al. Guideline for the management of nosocomial infections in South Africa. *South Afr J Epidemiol Infect.* 2006;21(3):152-160.
3. Recommendations for infection control for the practice of anaesthesiology. American Society of Anesthesiologists [homepage on the Internet]. 2012. Available from: <http://www.asahq.org/For-Members/Standards-Guidelines-and-Statements.aspx>
4. Lowman W, Venter L, Scribante J. Bacterial contamination of re-usable laryngoscope blades during the course of daily anaesthetic practice. *S Afr Med J.* 2013;103(6):386-389.
5. Samuel RA, Gopalan PD, Coovadia Y, Samuel R. Infection control in anaesthesia in regional, tertiary and central hospitals in KwaZulu-Natal. Part 1: Unsafe injection practices among anaesthetists. *South Afr J Anaesth Analg.* 2013;19(1):68-70.
6. Samuel RA, Gopalan PD, Coovadia Y, Samuel R. Infection control in anaesthesia in regional, tertiary and central hospitals in KwaZulu-Natal. Part 2: Equipment contamination. *South Afr J Anaesth Analg.* 2013;19(3):146-151.
7. Samuel RA, Gopalan PD, Coovadia Y, Samuel R. Infection control in anaesthesia in regional, tertiary and central hospitals in KwaZulu-Natal. Part 3: Decontamination practices. *South Afr J Anaesth Analg.* 2013;19(4):204-211.
8. Mphahlele MJ, Lukhwareni A, Burnett RJ, et al. High risk of occult hepatitis B virus infection in HIV-positive patients from South Africa. *J Clin Virol.* 2006;35(1):14-20.
9. Association of Anaesthetists of Great Britain and Ireland. Infection control in anaesthesia. *Anaesthesia.* 2008;63(9):1027-1036.
10. McQuoid-Mason D. Hospital-acquired infections: when are hospitals legally liable? *S Afr Med J.* 2012;102(6):353-354.



2. Executive Summary

Executive Summary: Part 1

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. 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 are 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.

Executive summary: Part 2

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.
- 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 check valve (one-way valve or non-return valve) in the infusion set does not prevent blood contamination of syringes or needles.

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 to dry 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.
- 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.



- 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 with alcohol before 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.

Executive Summary: Part 3

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 near the patient.
 - After removing gloves.
 - Before eating or drinking.
 - After using the bathroom.
- Plain soap (non-antimicrobial): used for routine hand washing, when hands look dirty. It will only remove loosely adherent transient bacteria, and will not decontaminate the hands.
- Antimicrobial soap: used if there has been visible hand contamination with blood or body fluids.
- 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 virtually no activity against bacterial spores and protozoal oocytes. Physical hand washing with water-based 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.
- 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.
- Gloves cannot be a substitute for hand washing as there always will be a measurable degree of glove leakage during use and self-contamination when removing the gloves.
- Bare below the elbow: False nails, nail polish, wrist watches and stoned rings must not be worn. Garments such as short sleeved or roll-up or push-up sleeves must be worn when in direct patient contact or performing hand washing.

Executive summary: Part 4

Anaesthetic equipment decontamination

Laryngoscope blades

Options for the reprocessing 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.
- 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 HLD is used, 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

Decontaminate after each patient.

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).

Decontaminate by sterilisation or HLD or intermediate-level disinfection (ILD).

Sterilisation:

- 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 battery removal.
- The manufacturer should be consulted to determine compatibility with the type of sterilisation. If the handle is not sterilisation friendly, it should be replaced.

High-level disinfection:

- 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, and which is well understood.
- HLD must be monitored and audited for compliance.
- Adequate numbers of laryngoscope handles per OT must be acquired to allow for battery removal.

Intermediate-level disinfection:

- ILD should take place after each patient.
- Chlorhexidine 2%/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.

Magill forceps

- Steam sterilise after each use.
- Adequate numbers of Magill forceps per OT should be acquired to allow for steam sterilisation.

Nasopharyngeal and rectal temperature probes

- Sterilise after each use according to manufacturers' recommendations.
- Adequate numbers of nasopharyngeal temperature probes per OT should be acquired to accommodate sterilisation.

Suction bowl

- Replace after each patient (one suction bowl per patient). The contaminated receiver should be sent for sterilisation.

Suction tubing

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

Oropharyngeal airways

- Single-patient use only.
- Discard 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

Breathing filters and breathing circuits

- 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 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.
- 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.
- 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 99.97% efficiency at a flow rate of 30 l/minute.
- The HMEF should be able to withstand a pressure of 60 hectopascals ($\approx 60 \text{ cmH}_2\text{O}$) without allowing liquid to pass through, or 20 hectopascals above the set pressure limit of the breathing circuit.



- 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 patients where there is a high risk of cross-infection as they do not prevent the passage of liquid though the filter. Electrostatic filters do not prevent transmission because liquid (carrying viruses and bacteria) 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.

The breathing circuit can be reused between cases, provided that:

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

The breathing circuit should be changed immediately:

- 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.

Bag valve mask resuscitators

Place a high-efficiency breathing filter between the valve and the mask before being used on a patient.

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

- 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 resuscitators should be sterilised:

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



Executive summary: Part 5

Prevention of intravascular catheter-related infection

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.
- Use ultrasound when possible.
- 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.
- All lines that are no longer needed should be removed promptly.

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.
- Maximal sterile barrier precautions to be used include the use of a cap, mask, sterile gown, sterile gloves and a sterile full body drape.
- The skin must be prepared with a solution that contains more than 0.5% chlorhexidine in 70% alcohol. If there is a contraindication to the use of chlorhexidine, tincture of iodine, an iodophore or 70% alcohol may be used as alternatives.
- The skin antiseptic must be allowed to dry prior to performing the procedure.

Catheter dressing and site management

- A sterile transparent semipermeable dressing must be used to cover the site. Sterile gauze 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 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.
- 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 for any pain, redness or purulence that may be suggestive of local infection.

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.
- Antimicrobial-impregnated catheters or those with antimicrobial properties may be considered in environments in which the rate of catheter-related bloodstream infections (CLABSI) is not decreasing, and lines are likely to remain in place for more than five days.

Management of lines and administration sets

- Any fat-containing emulsion (nutritional lipid emulsions or drugs with a fat emulsion-based vehicle) infusion sets in continuous use do not need to be changed more often than four days, but at least every seven days for patients not receiving blood or blood products.
- Infusion sets that have contained blood or blood products or fat-containing emulsions must be changed within 24 hours of initiating the infusion.
- 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.
- Needleless connectors and access ports on administration sets must be cleaned with 70% alcohol, tincture of iodine or chlorhexidine, prior to injection or connection.



Executive summary: Part 6

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 chlorhexidine with alcohol, should be used for skin preparation, and adequate time allowed for drying.
 - 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 is clinically necessary.

Peripheral nerve blocks

- Maximal barrier precautions are generally not necessary.
- Maximum barrier precautions should be used only 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.

Use of ultrasound

- A sterile probe and handle covering should be used, e.g. a sterile transducer sheath.
- The ultrasound machine and probes should be decontaminated before and after use, e.g. the ultrasound machine and probe should be wiped with a single-use towel to remove visible soiling, and then wiped with another single-use towel that has been soaked in an appropriate disinfectant, e.g. 70% isopropyl alcohol, and then allowed to dry.
- 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 gel sachet.



Executive summary: Part 7

Prevention of surgical site infection for the anaesthetist

Antibiotic prophylaxis

- Antibiotic prophylaxis should be administered before:
 - "Clean" surgery involving the placement of a prosthesis or implants.
 - "Clean-contaminated" surgery.
 - "Contaminated" surgery.
 - Do not give antibiotic prophylaxis for clean, non-prosthetic uncomplicated surgery.
- Give a single dose of antibiotic prophylaxis intravenously on starting anaesthesia 30 minutes before skin incision, but not more than one hour before incision. However, give antibiotic prophylaxis earlier for operations in which a tourniquet is used.
- A second dose of an antibiotic with a relatively short half-life, e.g. cephazolin, is often recommended for prolonged procedures.

- Consider antibiotic treatment in patients who have undergone surgery on a dirty or infected wound.

Physiological parameter homeostasis to reduce the risk of surgical site infections

- Maintain normoxia throughout the perioperative period.
- Give supplemental oxygen to maintain arterial saturation above 95%.
- Optimise the components that relate to oxygen delivery, i.e. haemoglobin levels, fractional inspiration of oxygen and cardiac output.
- Maintain optimal perfusion to the operative site.
- Avoid hypothermia.
- Avoid severe hyperglycaemia.
- Minimise blood transfusion and use leucodepleted blood in high-risk patients.



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.¹
- 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.²
- 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.³
- 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 obstetrics delivery suite and neonatal intensive care units (ICUs).
- 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 are 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.⁴

References

1. Association of Anaesthetists of Great Britain and Ireland. Infection control in anaesthesia. *Anaesthesia*. 2008; 63(9):1027-1036.
2. Samuel RA, Gopalan PD, Coovadia Y, Samuel R. Infection control in anaesthesia in regional, tertiary and central hospitals in KwaZulu- Natal. Part 3: Decontamination practices. *South Afr J Anaesth Analg*. 2013;19(4):204-211.
3. Samuel RA, Gopalan PD, Coovadia Y, Samuel R. Infection control in anaesthesia in regional, tertiary and central hospitals in KwaZulu-Natal. Part 1: Unsafe injection practices among anaesthetists. *South Afr J Anaesth Analg*. 2013;19(1):68-70.
4. Rowley E, Dingwall R. The use of single-use devices in anaesthesia: balancing the risks to patient safety. *Anaesthesia*. 2007;62(6):569-574.

Definitions and classifications

Table 1: 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	A process of removing organic and inorganic material from an object or surface with water and enzymatic products or detergent. It is the first step in all decontamination processes.
Disinfection	<p>A process that eliminates many or all pathogenic microorganisms, except bacterial spores, on objects or surfaces. There are three levels of disinfection based on the antimicrobial spectrum and rapidity of action:</p> <p>High-level disinfection (HLD)</p> <ul style="list-style-type: none"> • Destroys all microorganisms (mycobacteria, vegetative bacteria, viruses and fungal spores), except large numbers of bacterial spores in a relatively short exposure time. • <i>Examples of disinfectants:</i> Glutaraldehyde, orthophthalaldehyde, hydrogen peroxide and peracetic acid. • Used for semi-critical instrument decontamination. <p>Intermediate-level disinfection (ILD)</p> <ul style="list-style-type: none"> • Destroys mycobacteria, vegetative bacteria, most viruses, and most fungi, but does not kill bacterial spores. • <i>Examples of disinfectants:</i> 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. <p>Low-level disinfection (LLD)</p> <ul style="list-style-type: none"> • Destroys lipid or medium-sized viruses, some fungal spores and vegetative bacteria. • <i>Examples of disinfectants:</i> 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	<p>A process whereby all types of microorganisms, e.g. mycobacteria, vegetative bacteria, viruses and fungal spores, including bacterial endospores, are eliminated.</p> <p>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.</p>

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4. Safe injection practices and preventing the contamination of medication and fluids

4.1 Needles and syringes

Needles and syringes are sterile items, intended for single-patient use only.^{1,4}

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.^{1,4}

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.^{1,4}

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.^{1,2,4}

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.^{1,2,4}

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.^{2,4,5}

Before use, prepared syringes should be capped to avoid contamination.^{3,4}

After use or at the end of the anaesthetic, used syringes and needles should be discarded appropriately.³

Syringes must never be stored nor transported in clothing or pockets.²

The presence of a check valve (one-way valve or non-return valve) in the infusion set does not prevent the blood contamination of syringes or needles.^{4,5}

4.2 Preservative-free (single-dose) ampoules or vials

Preservative-free (single-dose) ampoules or vials are single-dose, single-patient items.^{3,4}

Do not give drugs from preservative-free vials or ampoules to multiple patients or save the remaining contents for later use.^{1,3,4}

Use of single-dose vials is preferred whenever possible over the use of multi-dose vials for parenteral medications.^{1,3,4}

They must be disposed after the drug dose has been drawn up and not reused for other patients.^{3,4}

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.⁴

4.3 Multi-dose vials

Use of single-dose vials is preferred whenever possible over that of multi-dose vials for parenteral medications.^{3,4}

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.⁴ A new sterile needle and syringe must be used each time the vial is entered.¹ 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.^{1,4} It must be discarded if there is suspicion that sterility has been compromised.^{1,4} 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.²

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

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.^{3,4}

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.^{1,2,4}

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.²

Use single-dose, single-use containers for flush solutions.²

Aseptic techniques should be used when preparing infusions, and breaks or taps in the lines kept to a minimum.³

Always clean IV injection ports with alcohol before use. Cover with a sterile cap after use.^{3,4}

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.⁴

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.⁴

Rationale

The silent and insidious, yet devastating, epidemic of unsafe injection practice is plaguing healthcare systems worldwide.⁶



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.⁷ Approximately 260 000 HIV/acquired immune deficiency syndrome (AIDS) cases, 2-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.⁷ 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.² 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.⁸ 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.⁹

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

Changing a needle (or cannula) and reusing the syringe is extremely dangerous.⁴ Negative pressure is generated when a needle is removed from a syringe, producing a siphoning effect that aspirates the needle contents into the syringe.⁵

IV administration tubing becomes contaminated with blood when backflow occurs during blood sample aspiration, or by accidental gravitation or from a blood transfusion.⁵ 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.^{4,5}

Infectious blood-borne organisms may be present, even if blood is not visible in the tubing.⁵ Injection at the most distal port from the IV cannula does not prevent contamination of the syringe.⁵

Single-use fentanyl ampoules should not be used on multiple patients undergoing spinal anaesthesia. Infectious complications of spinal anaesthesia include, but are not limited to, meningitis, encephalitis, and spinal and epidural abscesses.¹⁰ Potential pathogens, such as bacteria from airborne contaminants, non-

sterile glass fragments, or failure to use an aseptic technique, may contaminate open, partially used ampoules.¹¹

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.^{12,13} Viable viruses were found to survive for at least 24 hours in one study, while bacteria and endotoxin were also found in contaminated multi-dose vials.¹⁴

Injection ports are a route of entry for microorganisms into the vascular system or other sterile body compartments.¹⁵

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.¹⁶

Bacterial and fungal infections in post-surgical patients have been linked to contaminated infusions used on multiple patients.⁴ Propofol carries a high risk of contamination and cases of postsurgical sepsis and deaths from contaminated infusions have been reported.¹⁰ Propofol is manufactured in a nutrient-rich emulsion, containing glycerin, soy bean oil and egg phospholipids, and bacterial growth has been documented at six hours, despite the use of preservative.¹⁷ Therefore, manufacturers have recommended that propofol is discarded six hours after ampoule opening, and for continuous IV infusions, the tubing and any unused propofol after 12 hours.¹⁸

References

1. Guideline for isolation precautions: preventing transmission of infectious agents in healthcare settings. Centers for Disease Control and Prevention. CDC [homepage on the Internet]. 2007. <http://www.cdc.gov/ncidod/dhqp/pdf/isolation2007.pdf>
2. Dolan SA, Felizardo G, Barnes S, et al. APIC position paper: safe injection, infusion, and medication vial practices in health care. *Am J Infect Control*. 2010; 38(3):167-172.
3. The Association of Anaesthetists of Great Britain and Ireland. Infection control in anaesthesia. *Anaesthesia*. 2008;63(9):1027-1036.
4. American Society of Anaesthesiologists. Recommendations for infection control for the practice of anaesthesiology. 3rd ed. Boston: Little, Brown & Co; 1992.
5. Trepanier CA, Lessard MR, Brochu JG, Denault PH. Risk of cross infection related to the multiple use of disposable syringes. *Can J Anaesth*. 1990;37(2):156-159.
6. World Health Organization. SIGN Alliance Safe Injection Global Network: Advocacy booklet. WHO [homepage on the Internet]. 2011. Available from: http://www.who.int/injection_safety/sign/advocacy_booklet/en/index.html
7. Hauri AM, Armstrong GL, Hutin YJ. The global burden of disease attributable to contaminated injections given in health care settings. *Int J STD AIDS*. 2004;15(1):7-16.
8. Reid S, Van Niekerk AA. Injection risks and HIV transmission in the Republic of South Africa. *Int J STD AIDS*. 2009;20(12):816-819.
9. Samuel RA, Gopalan PD, Coovadia Y, Samuel R. Infection control in anaesthesia in regional, tertiary and central hospitals in KwaZulu-Natal. Part 1: Unsafe injection practices among anaesthetists. *South Afr J Anaesth Analg*. 2013;19(1):68-70.
10. American Society of Anesthesiologists. Practice advisory for the prevention, diagnosis and management of infectious complications associated with neuraxial techniques: a report by the American Society of Anesthesiologists Task Force on infectious complications associated with neuraxial techniques. *Anesthesiology*. 2010;112(3):530-545.



11. Zacher AN, Zornow MH, Evans G. Drug contamination from opening glass ampules. *Anesthesiology*. 1991;75(5):893-895.
12. Alter MJ, Ahtone J, Maynard JE. Hepatitis B virus transmission associated with a multiple-dose vial in haemodialysis unit. *Ann Intern Med*. 1983;99(3):330-333.
13. Mattner F, Gastmeier P. Bacterial contamination of multiple-dose vials: a prevalence study. *Am J Infect Control*. 2004;32(1):12-16.
14. Plott RT, Wagner RF, Tyring SK. Iatrogenic contamination of multidose vials in simulated use. A reassessment of current patient injection technique. *Arch Dermatol*. 1990;126(11):1441-1444.
15. O'Grady NP, Alexander M, Dellinger DP, et al. Guidelines for the prevention of intravascular catheter related infections. Centers for Disease Control and Prevention. *MMWR*. 2002;51(RR-10):1-29.
16. Macedo de Oliveira A, White KL, Leschinsky DP, et al. An outbreak of hepatitis C virus infections among outpatients at a hematology/ oncology clinic. *Ann Intern Med*. 2005;142(11):898-902.
17. Cole DC, Baslanti TO, Gravenstein NL, Gravenstein N. Leaving More than your fingerprint on the intravenous line: a prospective study on propofol anesthesia and implications of stopcock contamination. *Anesth Analg*. 2013. [Epub ahead of print].
18. Propofol Safety Signal. American Society of Anesthesiologists [homepage on the Internet]. 2007. Available from: <http://www.asahq.org/news/cdcalert053007.htm>



5. Hand hygiene guidelines

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.¹

Rationale

Our hands carry a high count [3.9×10^4 to 4.6×10^6 of colony-forming units (CFUs)/cm²] of resident and transient bacteria.^{2,6} 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.^{2,7}

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 10^6 skin squames per day,⁸ 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.^{2,9-17} In one study, 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.¹⁸ There was a 70% contamination rate for the healthcare workers who touched patients.¹⁸

Gloves do not fully protect against contamination.¹⁹ 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 Hayden¹⁸ on VRE contamination, 3% of VRE-negative healthcare workers who were wearing gloves were contaminated. This was 86% lower than their ungloved counterparts.¹⁸ Ehrenkranz et al¹⁹ 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 al²⁰ 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 faecalis*, *E. faecium*, *Pseudomonas aeruginosa* and *Burkholderia cepacia* when used in a study to show contamination by hand shaking. When the bacteria were suspended in saline, contamination occurred up to 30 min. However, if these bacteria were suspended in sputum, contamination occurred up to 180 minutes later.²¹

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.”²² 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.²³ Overall, between various studies, the compliance rate remained below 50%.²⁴⁻²⁶

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 al²⁰ 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 one per cent 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 1 ml of alcohol-based hand rub or liquid soap had lower bacterial reduction rates than 3 ml of the product.

Indications for hand hygiene are as follows:¹

- 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 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.²⁷⁻²⁹

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 rubs² 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.³⁰ Using inadequate volumes of alcohol-based rub (0.2-0.5ml) is as efficient as washing with plain soap and water.^{31,32} 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 12791²³ for hand rub formulations. Many commercially available hygienic rubs do not meet the standard EN 1500,³³ which is similar to EN 12791.

Alcohol has virtually no activity against bacterial spores and protozoal oozites.² 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^{2,5,34,35}

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.²

Gloves (sterile)

Sterile gloves must be worn if an invasive procedure is performed or if there is contact with sterile sites.

Gloves (non-sterile)

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. The need to be removed as soon as possible, and thus be changed between different procedures on 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 if there is inadequate time to perform hand washing, e.g. after intubation when the anaesthetic machine needs to be adjusted. Double gloves can also be worn. The outer glove may be removed before touching environmental surfaces are touched.

Gloves as a substitute for hand washing

Gloves cannot be a substitute for hand washing as there always will be a measurable degree of glove leakage during use and self-contamination when removing the gloves.

Powdered and polythene gloves should not be used.

Bare below the elbow

False nails, nail polish, wrist watches and stoned rings must not be worn. Garments such as short sleeved or roll-up or push-up sleeves must be worn when in direct patient contact or performing hand washing.

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. A direct correlation exists between these surfaces and stopcock contamination. Patients with positive stopcock cultures have a high incidence of post-surgical infections and mortality.

Anaesthetist workwear

Clothing should not impede effective hand washing, and should not come into contact with patients or environmental surfaces. Thus, the "bare below elbow" dictum must be followed.

Hand washing technique (three-stage technique)

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 that causes skin damage.
- *Stage 2: Washing and rinsing:* Rub hands together vigorously for 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.^{2,5}

References

1. American Society of Anesthesiologists. Recommendations for infection control for the practice of anaesthesiology. 3rd. ASA Committee on Occupational Health Task Force on Infection Control.
2. World Health Organization guidelines on hand hygiene in health care. Geneva: WHO; 2009.
3. Price PB. The bacteriology of normal skin: a new quantitative test applied to a



- study of the bacterial flora and the disinfectant action of mechanical cleansing. *J Infect Dis.* 1938;63:301-318.
4. Larson E. Effects of handwashing agent, handwashing frequency, and clinical area on hand flora. *Am J Infect Control.* 1984;12(2):76-82.
 5. Larson EL, Hughes CA, Pyrek JD, et al. Changes in bacterial flora associated with skin damage on hands of health care personnel. *Am J Infect Control.* 1998;26(5):513-521.
 6. Maki D. Control of colonization and transmission of pathogenic bacteria in the hospital. *Annals of Internal Medicine.* (1978;85 Pt 2 Suppl):777-780.
 7. Lark RL, van der Hyde K, Deeb GM, et al. An outbreak of coagulase- negative staphylococcal surgical-site infections following aortic valve replacement. *Infect Control Hosp Epidemiol.* 2001;22:618-623.
 8. Noble WC. Dispersal of skin microorganisms. *Br J Dermatol.* 1975;93(4):477-485.
 9. Pittet D, Dharan S, Touveneau S, et al. Bacterial contamination of the hands of hospital staff during routine patient care. *Arch Intern Med.* 1999;159(8):821-826.
 10. Samore MH, Venkataraman L, DeGirolami PC, et al. Clinical and molecular epidemiology of sporadic and clustered cases of nosocomial *Clostridium difficile* diarrhea. *Am J Med.* 1996;100(1):32-40.
 11. Boyce JM, Potter-Bynoe G, Chenevert C, King T. Environmental contamination due to methicillin-resistant *Staphylococcus aureus*: possible infection control implications. *Infect Control Hosp Epidemiol.* 1997;18:622-627.
 12. Ojajarvi J. Effectiveness of hand washing and disinfection methods in removing transient bacteria after patient nursing. *J Hyg (Lond).* 1980;85(2):193-203.
 13. Duckro AN, Blom DW, Lyle EA, et al. Transfer of vancomycin- resistant Enterococci via health care worker hands. *Arch Intern Med.* 2005;165(3):302-307.
 14. Lucet JC, Riquad MP, Mentre F, et al. Hand contamination before and after different hand hygiene techniques: a randomized clinical trial. *J Hosp Infect.* 2002;50(4):276-280.
 15. Bhalla A, Pultz NJ, Gries DM, et al. Acquisition of nosocomial pathogens on hands after contact with environmental surfaces near hospitalized patients. *Infect Control Hosp Epidemiol.* 2004;25(2):164-167.
 16. McBryde ES, Bradely LC, Whitby M, McElwain DL. An investigation of contact transmission of methicillin-resistant *Staphylococcus aureus*. *J Hosp Infect.* 2004;58(2):104-108.
 17. Ray AJ, Hoyen CK, Taub TF, et al. Nosocomial transmission of vancomycin-resistant enterococci from surfaces. *JAMA.* 2002;287(11):1400-1401.
 18. Hayden MK, Blom DW, Lyle EA, et al. Risk of hand or glove contamination after contact with patients colonized with vancomycin- resistant enterococcus or the colonized patients' environment. *Infect Control Hosp Epidemiol.* 2008;29(2):149-154.
 19. Pessoa-Silva CL, Dharan S, Huggonet S, et al. Dynamics of bacterial hand contamination during routine neonatal care. *Infect Control Hosp Epidemiol.* 2004;25(3):192-197.
 20. Noskin GA, Stosor V, Cooper I, Petersen LR. Recovery of vancomycin- resistant enterococci on fingertips and environmental surfaces. *Infect Control Hosp Epidemiol.* 1995;16(10):577-581.
 21. Doring G, et al. Distribution and transmission of *Pseudomonas aeruginosa* and *Burkholderia cepacia* in a hospital ward. *Pediatr Pulmonol.* 1996;21(2):90-100.
 22. Eckmanns T, Bessert J, Behnke M, et al. Compliance with antiseptic hand rub use in intensive care units: the Hawthorne effect. *Infect Control Hosp Epidemiol.* 2006;27(9):931-934.
 23. Rotter ML. European norms in hand hygiene. *J Hosp Infect.* 2004;56 Suppl 2:S6-S9.
 24. Pittet D, Mourouga P, Perneger TV. Compliance with handwashing in a teaching hospital. *Infection Control program.* *Ann Intern Med.* 1999;130(2):126-130.
 25. Nevin Kuzu, Fadime Özer, Semra Aydemir, et al. Compliance with hand hygiene and glove use in a university affiliated hospital. *Infect Control Hosp Epidemiol.* 2005;26(3): 312-315.
 26. Yawson AE, Hesse AA. Hand hygiene practices and resources in a teaching hospital in Ghana. *J Infect Dev Ctries.* 2013;7(4):338-347.
 27. Bottone EJ, Cheng M, Hymes S. Ineffectiveness of handwashing with lotion soap to remove nosocomial bacterial pathogens persisting on fingertips: a major link in their intrahospital spread. *Infect Control Hosp Epidemiol.* 2004;25(3):262-264.
 28. McFarland LV, Mulligan ME, Kwok RE, Stamm WE. Nosocomial acquisition of *Clostridium difficile* infection. *N Eng J Med.* 1989;320(4):204-210.
 29. Ehrenkranz NJ, Alfonso BC. Failure of bland soap handwash to prevent hand transfer of patient bacteria to urethral catheters. *Infect Control Hosp Epidemiol.* 1991;12(11):654-662.
 30. Kampf G, Ostermeyer C. Influence of applied volume on efficacy of 3-minute surgical reference disinfection method prEN 12791. *Appl Environ Microbiol.* 2004;70(12):7066-7069.
 31. Mackintosh CA, Hoffman PN. An extended model for transfer of micro- organisms via the hands: differences between organisms and the effect of alcohol disinfection. *Hyg (Lond).* 1984;92(2):345-355.
 32. Marples RR, Towers AG. A laboratory model for the investigation of contact transfer of micro-organisms. *J Hyg (Lond).* 1979;82:237-248.
 33. Kramer A, Rudolph P, Krampf G, Pittet D. Limited efficacy of alcohol- based hand gels. *Lancet.* 2002;359(9316):1489-1490.
 34. McNeil SA, Foster CL, Hedderwick SA, Kaufman CA. Effect of hand cleansing with antimicrobial soap or alcohol-based gel on microbial colonization of artificial fingernails worn by health care workers. *Clin Infect Dis.* 2001;32(3):367-372.
 35. Gupta A, Della-Latta P, Todd B, et al. Outbreak of extended-spectrum betalactamase- producing *Klebsiella pneumoniae* in a neonatal intensive care unit linked to artificial nails. *Infect Control Hosp Epidemiol.* 2004;25(3):210-215.



6. Anaesthetic equipment decontamination

6.1 Laryngoscopes

6.1.1 Laryngoscope blades

Contaminated anaesthetic equipment has been implicated in the nosocomial transmission of infectious diseases.¹

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 and which is complied with. 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.²⁻⁴ Other studies have identified RLBs as potential vectors for the transmission of methicillin-resistant *Staphylococcus aureus*, *P. aeruginosa*, *Serratia marcescens* and other pathogenic micro-organisms.^{5,6} Further studies have identified the presence of blood (occult and/or visible blood) on RLBs deemed to be ready for use.⁷⁻¹⁰ 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%.¹⁰ Staff from 11 of the 15 studied hospitals claimed that the blades were decontaminated by HLD.¹¹

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.¹² Hepatitis C has been shown to retain infectivity for several days at room temperature. Wenzel and Edmond acknowledged that instruments are sources of pulmonary infections with Gram- negative organisms, such

as *P. aeruginosa* or *S. marcescens*, reflecting an inanimate environmental reservoir.¹³ 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 on outbreaks of tuberculosis following bronchoscopic procedures.¹⁴

Concerns about high-level disinfection as a first-line method of decontamination of reusable laryngoscope blades in South Africa

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.^{15,16} 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.^{10,17} Bleeding in the mouth following routine laryngoscopy has been well described, as well as the contamination of laryngoscope blades with this blood.¹⁸ 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.¹⁹ 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.¹⁹ 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.^{17,20}

The rigidity of the high-level disinfection protocol

There are three essential stages in HLD:^{21,22}

- **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. orthophthalaldehyde (Cidex OPA[®]) and glutaraldehyde (Cidex[®]). The duration of immersion should be in accordance with the manufacturer's recommendations.
- **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.^{21,22}

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.¹¹

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.²³ 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^{22,24}

Commonly used high-level disinfectants in South Africa include orthophthalaldehyde (Cidex OPA[®]) and glutaraldehyde (Cidex[®]).

Orthophthalaldehyde (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 and using 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 were also reported in healthcare workers who were 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 certain that RLBs are appropriately decontaminated, they should use disposable equipment.²⁵

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.²⁶ 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 singleuse metal blades were more efficient than reusable metal blades. Significantly fewer failed first attempts and there were fewer poor-grade laryngeal views.²⁷

A plastic DLB is reported to be less efficient than a metal reusable blade during rapid sequence induction.²⁸ This is owing to the increased flexibility of the blade. Jabre et al recommended that conventional RLBs should be available for difficult intubations, and plastic DLBs used for uncomplicated intubations.²⁸



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.²⁶

The use of DLBs removes concerns about decontamination, and particularly the human factors.

The case for sterilisation of reusable laryngoscope blades

Sterilisation of RLBS has significant advantages over HLD:

- **Reliability:**^{21,22} 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 fibre-optic laryngoscope blades have been shown to deteriorate with repeated steam sterilisation, eventually becoming less bright.²⁹⁻³¹

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.³² 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 (AGBI) recommends that the anaesthetist wear gloves during intubation, and place the used blade in a designated receptacle to prevent contamination of surfaces, pillows and drapes.¹⁷

References

1. Wilson AJ, Nayak S. Disinfection, sterilization and disposables. *Anaesth Intensive Care Med.* 2013;4(10): 423-427.
2. Cullen MM, Trail A, Robinson M, et al. *Serratia marcescens* outbreak in a neonatal intensive care unit prompting review of decontamination of laryngoscopes. *J Hosp Infect.* 2005;59(1):68-70.
3. Nelson KE, Warren D, Tomasi AM, et al. Transmission of neonatal listeriosis in a delivery room. *Am J Dis Children.* 1985;139(9):903-905.
4. Foweraker JE. The laryngoscope as a potential source of cross-infection. *J Hosp Infect.* 1995;29(4):315-316.
5. Lowman W, Venter L, Scribante J. Bacterial contamination of re-usable laryngoscope blades during the course of daily anaesthetic practice. *S Afr Med J.* 2013;103(6):386-389.
6. Beamer JER, Cox RA. MRSA contamination of a laryngoscope blade: a potential vector for cross infection. *Anaesthesia.* 1999;54(10):1010-1011.
7. Morell RC, Ririe D, James RL, et al. survey of laryngoscope contamination at a university and a community hospital. *Anesthesiology.* 1994;80(4):960-966.
8. Ballin MS, McCluskey A, Maxwell S, Spilsbury S. Contamination of laryngoscopes. *Anaesthesia.* 1999;54(11):1115-1116.
9. Phillips RA, Monaghan WP. Incidence of visible and occult blood on laryngoscope blades and handles. *AANA J.* 1997;65(3):241-246.
10. Samuel RA, Gopalan PD, Coovadia Y, Samuel R. Infection control in anaesthesia in regional, tertiary and central hospitals in KwaZulu-Natal. Part 2: Equipment contamination. *S Afr J Anaesth Analg.* 2013;19(3):146-151.
11. Samuel RA, Gopalan PD, Coovadia Y, Samuel R. Infection control in anaesthesia in regional, tertiary and central hospitals in KwaZulu-Natal. Part 3: Decontamination practices. *S Afr J Anaesth Analg.* 2013;19(4):204-211.
12. Bond WW, Favero MS, Petersen NJ, et al. Survival of hepatitis B virus after drying and storage for one week. *Lancet.* 1981;317(8219):550-551.
13. Wenzel RP, Edmond MB. Tuberculosis infection after bronchoscopy. *JAMA.* 1997;278(13):1111.
14. Agerton T, Walway S, Gore B, et al. Transmission of highly drug-resistant strain (strain w1) of *Mycobacterium tuberculosis*: community outbreak and nosocomial transmission via a contaminated bronchoscope. *JAMA.* 1997;287(13):1073-1077.
15. Recommendations for infection control for the practice of anaesthesiology. American Society of Anesthesiologists [homepage on the Internet]. 2012. Available from: <http://www.asahq.org/For-Members/Standards-Guidelines-and-Statements.aspx>
16. Spaulding EH. Chemical disinfection of medical and surgical materials. In: Lawrence CA, Block SS, editors. *Disinfection, sterilization and preservation.* Philadelphia: Lea & Febiger, 1968; p.517-531.
17. Association of Anaesthetists of Great Britain and Ireland. Infection control in anaesthesia. *Anaesthesia.* 2008; 63(9):1027-1036.
18. Chrisco JA, Devane G. A descriptive study of blood in the mouth following routine oral endotracheal intubation. *J Am Assoc Nurse Anesthetists.* 1992;60:379-383.
19. Panlilio AL, Cardo DM, Grohskopf LA, et al. Updated US Public Health Service guidelines for the management of occupational exposures to HIV and recommendations for postexposure prophylaxis. *MMWR Recomm Rep.* 2005;54(RR-9):1-17.
20. Dutch Working Party on Infection Prevention. Cleaning and disinfection of endoscopes. Number 21b. 2001.
21. Muscarella LF. Prevention of disease transmission during flexible laryngoscopy. *Am J Infect Control.* 2007;35(8):536-544.
22. Guideline for disinfection and sterilization in healthcare facilities. Centers for Disease Control and Prevention [homepage on the Internet]. 2008. Available from: http://www.cdc.gov/hicpac/Disinfection_Sterilization/toc.html
23. Muscarella LF. Recommendations to resolve inconsistent guidelines for reprocessing of sheathed and unsheathed rigid laryngoscopes. *Infect Control Hosp Epidemiol.* 2007;28(4):504-507.
24. Occupational Safety and Health Administration. Best practices for the safe use of glutaraldehyde in health care. US Department of Labor; 2006.
25. Machan MD, Monaghan WP, McDonough J, et al. Emerging evidence in infection



control: effecting change regarding use of disposable laryngoscope blades. AANA Journal. 2013;81(2):103-108.

26. Rowley E, Dingwall R. The use of single-use devices in anaesthesia: balancing the risks to patient safety. *Anaesthesia*. 2007;62(6):569-574.
27. Amour J, Manach YL, Borel M, et al. Comparison of single-use and reusable metal laryngoscope blades for orotracheal intubation during rapid sequence induction of anesthesia: a multicentre cluster randomized study. *Anesthesiology*. 2010;112(2):325-332.
28. Jabre P, Leroux B, Brohon S, et al. A comparison of plastic single-use with metallic reusable laryngoscope blades for out-of-hospital tracheal intubation. *Ann Emerg Med*. 2007;50(3):258-263.
29. Bucx MJ, Veldman DJ, Beenhakker MM, Koster R. The effect of steam sterilisation at 134 C on light intensity provided by fibrelight Macintosh laryngoscopes. *Anaesthesia*. 1999;54(9):875-878.
30. Bucx MJL, Gast M, Veldhuis J, Hassing LH, et al. The effect of mechanical cleaning and thermal disinfection on light intensity provided by fibrelight Macintosh laryngoscopes. *Anaesthesia*. 2003;58(5):461-465.
31. Nishiyama T. Changes in the light intensity of the fiberoptic laryngoscope blade by steam sterilization. *Anesth Analg*. 2007;104(4):908-910.
32. Gadalla F, Fong J. Improved infection control in the operating room. *Anesthesiology*. 1990;73(6):1295.

6.1.2 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. (This does not change the need for HLD or sterilisation). Consider using the specifically devised double-glove, clean induction technique by Gadalla and Fong.

Decontaminate by sterilisation, or HLD or intermediate-level disinfection (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. 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, and which is well 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%/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.

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.¹ 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.²⁻⁴ 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.^{3,5-7}

Traditionally, laryngoscope handles have a knurled surface for good grip. However, the fissures in this surface may provide a protected niche for pathogens.^{2,8} 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.⁷ 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.^{7,9}

Williams et al 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.⁷ 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.¹⁰

Choice of strategies to prevent cross-infection will be influenced by a cost-benefit analysis, but include the use of newly sterilised laryngoscope handles for each case, disposable laryngoscope handles or HLD of handles between each case.¹ ILD with



chlorhexidine 2%/alcohol 70% has been used.¹¹ 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.¹¹

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.^{9,12}

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.¹³ It is imperative that laryngoscope handles are decontaminated between use.^{12,14}

References

- Negri de Sousa AC, Levy CE, Freitas MI. Laryngoscope blades and handles as sources of cross-infection: an integrative review. *J Hosp Infect.* 2013;83(4):269-275.
- Esler MD, Baines LC, Wilkinson DJ, Langford RM. Decontamination of laryngoscopes: a survey of national practice. *Anaesthesia.* 1999;54(6):587-592.
- Simmons SA. Laryngoscope handles: a potential for infection. *AANA J.* 2000;68(3):233-236.
- Morell RC, Ririe D, James RL, et al. Survey of laryngoscope contamination at a university and a community hospital. *Anesthesiology.* 1994;80:960-966.
- Ballin MS, McCluskey A, Maxwell S, Spilsbury S. Contamination of laryngoscopes. *Anaesthesia.* 1999;54(11):1115-1116.
- Call TR, Auerbach FJ, Riddell SW, et al. Nosocomial contamination of laryngoscope handles: challenging current guidelines. *Anesth Analg.* 2009;109(2):479-483.
- Williams D, Dingley J, Jones C, Berry N. Contamination of laryngoscope handles. *J Hosp Infect.* 2010;74(2):123-128.
- Yee KF. Decontamination issues and perceived reliability of the laryngoscope: a clinician's perspective. *Anaesth Intensive Care.* 2003;31(6):658-662.
- Muscarella LF. Recommendations to resolve inconsistent guidelines for the reprocessing of sheathed and unsheathed rigid laryngoscopes. *Infect Control Hosp Epidemiol.* 2007;28(4):504-507.
- Medical device alert: reusable laryngoscope handles. Medicines and Healthcare Products Regulatory Agency [homepage on the Internet]. 2011. Available from: <http://www.mhra.gov.uk/Publications/Safetywarnings/MedicalDeviceAlerts/CON129213>
- Howell V, Thoppil A, Young H, et al. Chlorhexidine to maintain cleanliness of laryngoscope handles. An audit and laboratory study. *Eur J Anaesthesiol.* 2013;30(5):216-221.
- The Association of Anaesthetists of Great Britain and Ireland. Infection control in anaesthesia 2. *Anaesthesia.* 2008;63(9):1027-1036.

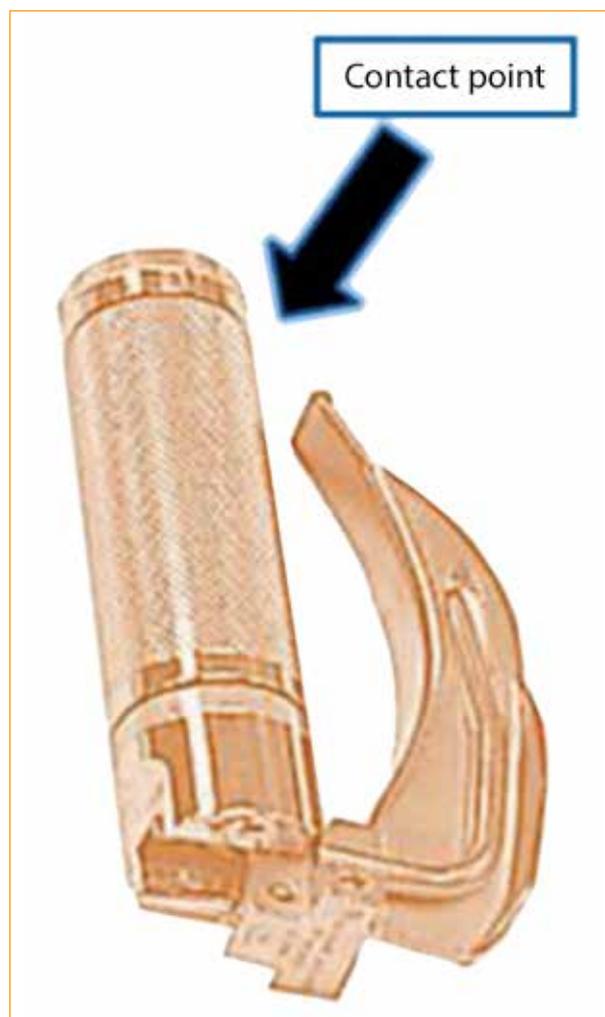


Figure 1: Contact point between the laryngoscope blade and the handle*

* Reproduced with permission¹³

- Samuel RA, Gopalan PD, Coovadia Y, Samuel R. Infection control in anaesthesia in regional, tertiary and central hospitals in KwaZulu-Natal. Part 2: Equipment contamination. *S Afr J Anaesth Analg.* 2013; 19(3):146-151.
- Guidelines on infection control in anaesthesia. Australian and New Zealand Colleges of Anaesthetists; 2005.



Step-by-step decontamination of laryngoscope blades and handles

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 stabilisation. Therefore avoid delays in reprocessing after use.

Step 2: Disassembly in the decontamination area

Detach the laryngoscope blade from the handle.

Remove the fibre-optic 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.

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: i.e. orthophthalaldehyde (Cidex OPA[®]) and glutaraldehyde (Cidex[®]). The duration of immersion should be in accordance with the manufacturer's recommendations.

After HLD, rinse the blade and/or handle with 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 storage area, using care to prevent recontamination.

The storage area should be clean and dry.

To avoid bacterial colonization, the blade and handle should not be stored in a closed carrying case, container or kit.



6.2 Bronchoscopes

Step-by-step decontamination of 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.

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 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 prevents 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 sterilization.

Unless otherwise recommended by the manufacturer, the bronchoscope should be completely immersed in a freshly made solution of water and low-sudsing 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 laryngoscope.

Ensure that all necessary personal protective equipment 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 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 steam 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 endoscopy 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 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.



6.3 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.

6.4 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.

6.5 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.¹ 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.

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.²

Sterilisation is recommended as it has a large margin of safety, coupled with reliability, consistency and lethality.³ It also removes the "human" element from the process of decontamination, and is relatively inexpensive.

References

1. Samuel RA, Gopalan PD, Coovadia Y, Samuel R. Infection control in anaesthesia in regional, tertiary and central hospitals in KwaZulu-Natal. Part 2: Equipment contamination. *S Afr J Anaesth Analg*. 2013;19(3):146-151.
2. Samuel RA, Gopalan PD, Coovadia Y, Samuel R. Infection control in anaesthesia in regional, tertiary and central hospitals in KwaZulu-Natal. Part 3: Decontamination practices. *S Afr J Anaesth Analg*. 2013;19(4):204-211.
3. Guideline for disinfection and sterilization in healthcare facilities. Centers for Disease Control and Prevention [homepage on the Internet]. 2008. Available from: http://www.cdc.gov/hicpac/Disinfection_Sterilization/toc.html

6.6 Suction tubing

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

6.7 Oropharyngeal airways

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

6.8 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.¹

Alternative single-use intubation aids are preferable to bougie use. Intubation aids and stylets are single-use items.

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.²

It has been noted that bougies, and intubation guides and stylets have become contaminated with pathogenic bacteria³ after reuse, and have been associated with cross-infection.⁴

References

1. Association of Anaesthetists of Great Britain and Ireland. Infection control in anaesthesia. *Anaesthesia*. 2008;63(9):1027-1036.
2. Miller DH, Youkhana I, Karunaratne WU, Pearce A. Presence of protein deposits on cleaned re-usable anaesthetic equipment. *Anaesthesia*. 2001;56(11):1069-1072.
3. Jerwood DC, Mortiboy D. Disinfection of gum elastic bougies. *Anaesthesia*. 1995;50(4):376.
4. Sriparkdeea C, Rujirojindakula P, Pakpiroma J, et al. Contamination of intubating stylets in operating rooms. *J Hosp Infect*. 2010;75:325-337.

6.9 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.^{1,2}

- The filter should be placed on the Y-piece between the endotracheal or tracheostomy tube and the elbow connector or breathing circuit.^{3,4-7}
- 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.⁸
- 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.⁹⁻¹⁹
- 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.^{1,20-23}
- The filter has to be changed when it becomes visibly contaminated with blood or secretions, or with condensate within the breathing system.²⁴

6.9.1 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 EN13328-1.²⁵

The HMEF should have a 99.97% efficiency at a flow rate of 30 l/minute.²⁵

The HMEF should be able to withstand a pressure of 60 hectopascals (≈ 60 cmH₂O) without allowing liquid to pass through, or 20 hectopascals above the set pressure limit of the breathing circuit.^{3,26}

The HMEF must have a minimum humidity output of 20 g/m³ in patients ventilated < 10 hours or 33 g/m³ in ICU patients ventilated > 10 hours.^{25,27,28}

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.³

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.^{3,8,29-34} 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.^{24,35} The lower-weight limit should be $\leq 24,35$ a heat- and moisture exchanger (HME) of 5 kg and filter of 3 kg.

6.9.2 Breathing circuits

The breathing system consists of the elbow connector or catheter mount, the breathing circuit, the reservoir bag and CO₂ absorber.

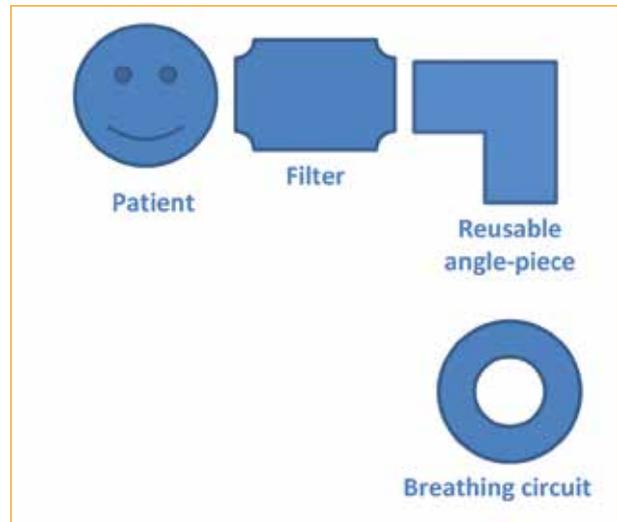


Figure 2: Correct placement of filter

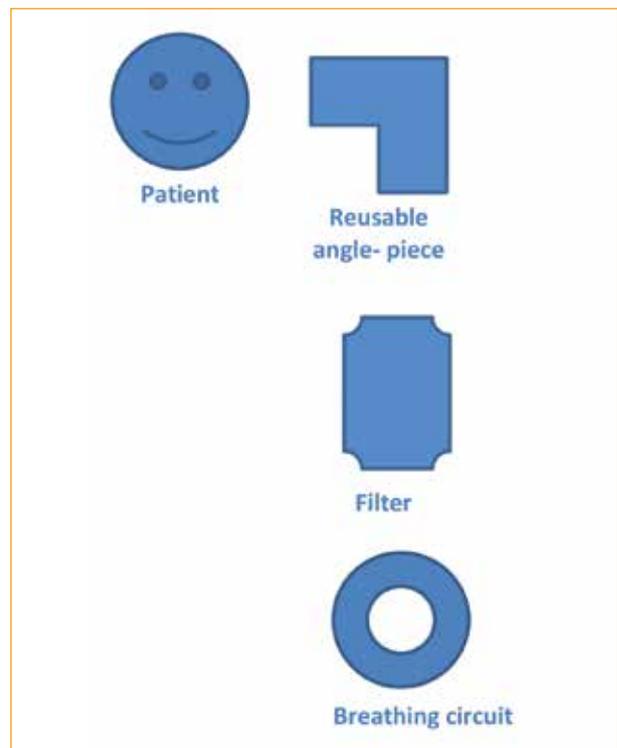


Figure 3: Incorrect placement of filter

The components of the breathing circuit can be re-used 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.^{36,37}

The breathing system components are seen as semi-critical items, and should be disinfected according to the manufacturer's instructions.^{5,38,39}



The CO₂-absorber canister should be cleaned every time the absorber material is changed. Disinfection must take place according to manufacturer's guidelines.^{38,40}

The components of the breathing circuit should be changed immediately in any of the following circumstances:⁴¹

- 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.

6.9.3 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.⁴²

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^{40,43} *A. calcoaceticus*, *P. maltophilia*, *P. aeruginosa*, *Flavobacterium meningosepticum*, *K. pneumoniae*, *P. mirabilis*, *Enterobacter cloacae*, *Citrobacter diversus*, *E. agglomerans*, *Candida albicans*, *E. cloacae*, *Proteus* spp. and *Streptococcus* spp. 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 re-use of anaesthetic breathing circuits between patients in a cost-constrained environment.

Another benefit of combining a viral or bacterial filter with a heat-and-moisture-exchange 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.^{25,26,44}

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 heat-and-moisture exchange. 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.²⁵

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.^{25,45} 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.^{24,45}

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 demonstrated that up to 5.6% of contamination occurs in breathing circuits that had been in use for 72 hours with electrostatic filters.²⁹

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.^{25,46}

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.²⁵ 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 re-used among patients.

Some studies have shown that filters placed at the Y-connection were able to prevent the contamination of breathing systems.⁴⁻⁷ It is also known that filters reduce airborne microbes when tested in vitro.⁴⁷⁻⁵² 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 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.⁵³



What is clear is the risk of complications associated with the use of filters.⁹⁻²³ 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 lyme, 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 societies² have taken a different stance in view of cost constraints, and in their guidelines have recommended that breathing circuits can be re-used, 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.^{3,26} The British Society for Antimicrobial Chemotherapy recommends the use of HMEs rather than heated humidifiers to reduce the incidence of ventilator-associated pneumonia.¹

When circuits are to be re-used, it is recommended that a high efficiency filter is used.² Chant et al⁵⁴ 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 were typed to be the same strain. A second such case was also reported.⁵⁵

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 re-using 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.⁵⁶ 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 re-used between patients. Ip et al⁴² 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 2003.⁵⁷

References

1. Masterton RG, Galloway A, French G, et al. Guidelines for the management of hospital-acquired pneumonia in the UK: report of the working party on hospital-acquired pneumonia of the British Society for Antimicrobial Chemotherapy. *J Antimicrob Chemother.* 2009;62(1):5-34.
2. European Society of Intensive Care Medicines Task Force for ICU triage. ESICM [homepage on the Internet]. Available from: <http://www.esicm.org/>
3. Wilkes AR. Heat and moisture exchangers and breathing system filters: their use in anaesthesia and intensive care. Part 1: History, principles and efficiency. *Anaesthesia.* 2011;66(1):31-39.
4. Vézina DP, Trépanier CA, Lessard MR, et al. Anesthesia breathing circuits protected by the DAR Barrierbac S breathing filter have a low bacterial contamination rate. *Can J Anesth.* 2001;48(8):748-752.
5. Vézina DP, Trépanier CA, Lessard MR, et al. An in vivo evaluation of the mycobacterial filtration efficacy of three breathing filters used in anesthesia. *Anesthesiology.* 2004;101(1):104-109.
6. Daggan R, Zefeiridis A, Steinberg D, et al. High-quality filtration allows reuse of anesthesia breathing circuits resulting in cost savings and reduced medical waste. *J Clin Anesth.* 1999;11(7):536-539.
7. Rathgeber J, Kietzmann D, Mergeryan H, et al. Prevention of patient bacterial contamination of anaesthesia-circle-systems: a clinical study of the contamination risk and performance of different heat and moisture exchangers with electret filter (HMEF). *Eur J Anaesthesiol.* 1997;14(4):368-373.
8. Wilkes AR. The ability of breathing system filters to prevent liquid contamination of breathing systems: a laboratory study. *Anaesthesia.* 2002;57(1):33-39.
9. Aarhus D, Soreide E, Holst-Larsen H. Mechanical obstruction in the anesthesia delivery system mimicking severe broncho-spasm. *Anaesthesia.* 1997;52(10):989-997.
10. Barnes S, Normoyle D. Failure of ventilation in an infant due to increased resistance of a disposable heat and moisture exchanger. *Anesth Analg.* 1996;83(1):193.
11. Buckley PM. Increase in resistance of in-line breathing filters in humidified air. *Br J Anaesth.* 1984;56(6): 637-643.
12. Villafane, MC, Cinnella G, Lofaso F, et al. Gradual reduction of endotracheal tube diameter during mechanical ventilation via different humidification devices. *Anesthesiology.* 1996;85(6):1341-1349.
13. Cohen IL, Weinberg PF, Fein IL, Rowinski GS. Endotracheal tube occlusion associated with the use of heat and moisture exchangers in the intensive care unit. *Crit Care Med.* 1988;16(3):277-279.
14. Barton RM. Detection of expiratory antibacterial filter occlusion. *Anesth Analg.* 1993;77(1):197.
15. Heat and moisture exchange filter obstruction. *Australian Therapeutic Device Bulletin;* 1990.
16. Prados W. A dangerous defect in a heat and moisture exchanger. *Anesthesiology.* 1989;71(5):804.
17. Prasad KK, Chen L. Complications related to the use of a heat and moisture exchanger. *Anesthesiology.* 1990;72(5):958.
18. Walton JS, Fears R, Burt N, Dorman HB. Intraoperative breathing circuit obstruction caused by Albuterol nebulization. *Anesth Analg.* 1999;89(3):650-651.
19. Williams DJ, Stacey MR. Rapid and complete occlusion of a heat and moisture exchange filter by pulmonary edema (clinical report). *Can J Anaesth.* 2002;49(2):126-131.
20. Moon RE. Cause of CO poisoning, relation to halogenated agents still not clear. *J Clin Monit.* 1995;11(1): 66-67.
21. Henriksson BA, Sundling J, Hellman A. The effect of a heat and moisture exchanger on humidity in a low-flow anaesthesia system. *Anaesthesia.* 1997;52(2):144-149.
22. Frink EJ Jr, Nogami WM, Morgan SE, Salmon RC. High carboxyhemoglobin concentrations occur in swine during desflurane anesthesia in the presence of partially dried carbon dioxide absorbents. *Anesthesiology.* 1997;87(2):308-316.



23. Lentz RE. CO poisoning during anaesthesia poses puzzle. *J Clin Monit*. 1995;11(1):67-71.
24. Wilkes AR. Heat and moisture exchangers and breathing system filters: their use in anaesthesia and intensive care. Part 2: Practical use, including problems, and their use with paediatric patients. *Anaesthesia*. 2011;66(1):40-51.
25. Lawes EG. Hidden hazards and dangers associated with the use of HME/filters in the breathing circuits. Their effect on toxic metabolite production, pulse oximetry and airway resistance. *Br J Anaesth*. 2003;91(2):249-264.
26. Kramer A, Kranabetter R, Rathgebe J, et al. Infection prevention during anaesthesia by the use of breathing system filters (BSF): joint recommendation by German Society of Hospital Hygiene and German Society for anaesthesiology and Intensive Care. *GMS Krankenhaushygiene Interdisziplinär*. 2010;5:1-19.
27. Chamney AR. Humidification requirements and techniques. Including a review of the performance of equipment in current use. *Anaesthesia*. 1969;24(4):602-617.
28. Kleeman PP. Humidity of anaesthetic gases with respect to low flow anaesthesia. *Anaesth Intensive Care*. 1994;22(4):396-408.
29. Hartmann D, Jung M, Neubert TR, et al. Microbiological risk of anaesthetic breathing circuits after extended use. *Acta Anaesthesiologica Scandinavica*. 2008;52(3):432-436.
30. Lee MG, Ford JL, Hunt PB, et al. Bacterial retention properties of heat and moisture exchange filters. *Br J Anaesth*. 1992;69(5):522-525.
31. Hedley RM, Allt-Graham J. A comparison of the filtration properties of heat and moisture exchangers. *Anaesthesia*. 1992;47(5):414-420.
32. Vandembroucke-Grauls CMJE, Teeuw KB, Ballemans K, et al. Bacterial and viral removal efficiency, heat and moisture exchange properties of four filtration devices. *J Hosp Infect*. 1995;29(1):45-56.
33. Cann C, Hampson MA, Wilkes AR, Hall JE. The pressure required to force liquid through breathing system filters. *Anaesthesia*. 2006;61(5):492-497.
34. Scott DHT, Fraser S, Willson P, et al. Passage of pathogenic microorganisms through breathing system filters used in anaesthesia and intensive care. *Anaesthesia*. 2010;65(7):670-673.
35. Carter JA. Filters and small infants. *Anaesthesia*. 2004;59(7):719-720.
36. Carter JA. The reuse of breathing systems in anaesthesia. *Respir Care Clin N Am*. 2006;12(2):275-286.
37. Hübner NO, Daeschlein G, Kobayashi H, et al. Microbiological safety and cost-effectiveness of weekly breathing circuit changes in combination with heat moisture exchange filters: a prospective longitudinal clinical survey. *J Infect Chemother*. [Epub ahead of print].
38. Juwarkar CS. Cleaning and sterilisation of anaesthetic equipment. *Indian J Anaesth*. 2013;57(5):541-550.
39. Standards Association. Decontamination of reusable medical devices. CSA; 2008.
40. Demers RR. Bacterial/viral filtration: let the breather beware! *Chest*. 2001;120(4):1377-1389.
41. Hübner NO, Daeschlein G, Kobayashi H, et al. Microbiological safety and cost-effectiveness of weekly breathing circuit changes in combination with heat moisture exchange filters: a prospective longitudinal clinical survey. *J Infect Chemother* [Epub ahead of print].
42. Ip M, Tang JW, Hui DS, et al. Airflow and droplet spreading around oxygen masks: a simulation control research. *Am J Infect Control*. 2007;35(10):684-689.
43. Malecka-Griggs B, Reinhardt DJ. Direct dilution sampling, quantitation, and microbial assessment of open system ventilation circuits in intensive care units. *J Clin Microbiol*. 1983;17(5):870-877.
44. Williams R, Rankin N, Smith T, et al. Relationship between humidity and temperature of inspired gas and the function of airway mucosa. *Crit Care Med*. 1996;24(11):1920-1929.
45. Wilkes AR. Breathing filters, humidifiers and nebulizers. In: Davey AJ, Diba A, editors. *Ward's anaesthetic equipment*. 5th ed. London: WB Saunders, 2005; p. 291-303.
46. ISO9360-1:2000. Anaesthetic and respiratory equipment heat and moisture exchangers (HMEs) for humidifying respired gasses in humans. Part 1: HMEs for use with minimum tidal volumes of 250 ml. ISO [homepage on the Internet]. Available from: www.iso.org
47. Mebius C. Heat and moisture exchangers with bacterial filters: a laboratory study. *Acta Anaesthesiologica Scandinavica*. 1992;36(6):572-576.
48. Holton J, Webb AR. An evaluation of the microbial retention performance of three ventilator-circuit filters. *Intensive Care Med*. 1994;20(3):233-237.
49. Lee MG, Ford JL, Hunt PB, et al. Bacterial retention properties of heat and moisture exchange filters. *Br J Anaesth*. 1992;69(5):522-525.
50. Hedley RM, Allt-Graham J. A comparison of the filtration properties of heat and moisture exchangers. *Anaesthesia*. 1992;47(5):414-420.
51. Vandembroucke-Grauls CMJE, Teeuw KB, Ballemans K, et al. Bacterial and viral removal efficiency, heat and moisture exchange properties of four filtration devices. *J Hosp Infect*. 1995;29(1):45-56.
52. Wilkes AR, Benbough JE, Speight SE, Harmer M. The bacterial and viral filtration performance of breathing system filters. *Anaesthesia*. 2000;55(5):458-465.
53. American Thoracic Society guidelines for the management of adults with hospital acquired ventilator associated and health care associated pneumonia. *Medicine*. 2005;171(4):388-416.
54. Chant K, Kociuba K, Munro R, et al. Investigation of possible patient-to-patient transmission of Hepatitis C in a hospital. *New South Wales Public Health Bulletin*. 1994;5:47-51.
55. Heinsen A, Bendtsen F, Fomsgaard A. A phylogenetic analysis elucidating a case of patient-to-patient transmission of hepatitis C virus during surgery. *J Hosp Infect*. 2000;46(4):309-313.
56. Sutcliffe AJ. Can the life of "single use" breathing systems be extended? *Anesthesia*. 1995;50(4):283-285.
57. Lee N, Hui DS, Wu A, et al. A major outbreak of severe acute respiratory syndrome in Hong Kong. *N Engl J Med*. 2003;348(20):1986-1994.

6.10 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.¹

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.^{2,3}

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.²

Do not disassemble the pressure release valve and the positive end-expiratory pressure valve.²

All of the parts should be rinsed in clean water to remove the detergent.²

All of the parts should be allowed to dry in a clean controlled environment, where the risk for recontamination is low.^{2,3}

The components should then be subjected to one of the following decontamination techniques:³

- 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:²

- 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.⁴ 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.^{2,3}

It has been shown that bacteria can be cultured from resuscitators which were macroscopically “clean”.⁵ Paediatric Ambu® bags in the obstetric OT are used more frequently than adult ones, and have been linked with disease transmission.

References

1. European Society of Intensive Care Medicines Task Force for ICU triage. ESICM [homepage on the Internet]. Available from: <http://www.ecism.org>
2. Instructions for the use of Silicone Manual Resuscitator [homepage on the Internet]. Available from: <http://www.promed.ie/shop/assets/catalog/parts/15224Megr%20Resus.pdf>
3. Directions for use: Laerdal Silicone Resuscitator. Laederal Medical [homepage on the Internet]. Available from: www.laerdal.com
4. Equipment decontamination and disinfection. National Institute of Health [homepage on the Internet]. Available from: <http://clinicalcenter.nih.gov/>
5. Bacteria lurking in resuscitation bags. Medscape [homepage on the Internet]. 2013. Available from: <http://www.medscape.com>

6.11 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 that 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.¹ Variant Creutzfeldt-Jakob disease, a new transmissible prion disease, was identified in humans in Great Britain in 1996.² Recently, large numbers of prion proteins were detected in human tonsillar tissue, raising significant concern in several countries about reusable anaesthetic and surgical equipment.³ 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.⁴⁻⁷ 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.⁸ 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.⁹
- *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.¹⁰
- *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

References

1. Hill AF, Desbruslais M, Joiner S, et al. The same prion strain causes CJD and BSE. *Nature*. 1997;389(6650): 448-450.
2. Will RG, Ironside JW, Zeidler M. A new variant of Creutzfeldt-Jacob disease in the UK. *Lancet*. 1999;347(9006):921-925.
3. Hill AF, Butterworth RJ, Joiner S, et al. Investigation of variant Creutzfeldt-Jakob disease and other human prion diseases with tonsil biopsy samples. *Lancet*. 1999;353(9148):183-189.
4. Clery G, Brimacombe J, Stone T, et al. Routine cleaning and autoclaving does not remove protein deposits from reusable laryngeal mask devices. *Anesth Analg* 2003;97(4):1189-1191.
5. Miller DM, Youkhana I, Karunaratne WU, et al. Presence of protein deposits on “cleaned” re-usable anaesthetic equipment. *Anaesthesia*. 2001;56(11):1069-1072.
6. Greenwood J, Green N, Power G. Protein contamination of the laryngeal mask airway and its relationship to re-use. *Anaesth Intensive Care*. 2006;34(3):343-346.
7. Chu LF, Mathur P, Trudell JR, Brock-Utne JG. Contamination problems with reuse of laryngeal mask airways and laryngoscopes. *Saudi J Anaesth*. 2008;2:58-61.
8. Cousens SN, Vynnycky E, Zeidler M, et al. Predicting the CJD epidemic in humans. *Nature*. 1997;385(6613): 197-198.
9. LMA™. 40 use program [homepage on the Internet]. Available from: <http://www.lmana.com/files/40usepackage-lma412.pdf?PHPSESSID=18748c2a4578e01bdff968c059acc04>
10. Rosenberg H, Marr A, Smith C. What is the cost of using an LMA? *Anesthesiol*. 2001;95:A506.
11. Rowley E, Dingwall R. The use of single-use devices in anaesthesia: balancing the risks to patient safety. *Anaesthesia*. 2007;62(2):569-574.



6.12 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.^{1,2} Loftus et al recently reported that multidrug-resistant bacterial transmission to the anaesthesia work area occurred during the practice of general anaesthesia.³

References

1. Perry SM, Monaghan WP. The prevalence of visible and/or occult blood on anesthesia and monitoring equipment. *AANA Journal*. 2001;69(1):44-49.
2. Albert NM, Hancock K, Murray T, et al. Cleaned, ready-to-use, reusable electrocardiographic lead wires as a source of pathogenic microorganisms. *Am J Crit Care*. 2010;19(6):73-80.
3. Loftus RW, Koft MD, Burchmann CC, et al. Transmission of pathogenic bacterial organisms in the anesthesia work area. *Anesthesiology*. 2008;109(3):399-407.



7. Prevention of intravascular catheter-related 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.

Evidence that simple interventions reduce the incidence of sepsis significantly is key to the implementation of these guidelines.¹ Institutions should be able to produce data on the incidence of catheter-related bloodstream infections (CLABSI) and promote care bundles and interventions that have been shown to reduce the incidence of CLABSI effectively.

7.1 The placement of central venous catheters

Recommendations

The subclavian site is preferred over either the internal jugular or femoral sites in adult patients in order to reduce the incidence of sepsis.²

Use ultrasound when possible and when trained operators are available to reduce the number of cannulation attempts and the incidence of mechanical complications.³

Use a line with the minimum number of lumens necessary to facilitate management of the patient.⁴

When adherence to sterile technique cannot be assured, the line must be removed as soon as possible, but within 48 hours.⁵

All lines that are no longer needed should be removed promptly.

Rationale

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 dry and not subject to the accumulation of sweat.

Ultrasound, in the hands of individuals who have been appropriately trained, has been shown to be effective in reducing cannulation attempts and complications.

Each individual lumen in a line adds to the risk of the development of sepsis. Lumens that are not actively running fluid carry an added risk of sepsis.

Non-adherence to sterile technique should not be condoned, and lines must not be placed in such circumstances unless immediately life-saving.

Lines that have been removed carry no risk of sepsis whatsoever.

7.2 Sterile technique for the placement of central venous catheters

Recommendations

The operator should scrub, as for a surgical procedure, prior to the placement of a central venous catheter.⁶

Maximal sterile barrier precautions to be used include the use of a cap, mask, sterile gown, sterile gloves and a sterile full body drape.⁷

The skin must be prepared with a solution that contains more than 0.5% chlorhexidine in 70% alcohol.⁸

If there is a contraindication to the use of chlorhexidine, tincture of iodine, an iodophore or 70% alcohol may be used as alternatives.⁹

The skin antiseptic must be allowed to dry prior to performance of the procedure.^{8,9}

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.

7.3 Catheter dressing and site management

Recommendations

A sterile transparent semi-permeable dressing must be used to cover the site. Sterile gauze may be used as an alternative.^{10,11}

Sterile gauze should be used if there is any bleeding, exudate or excessive skin moisture that accumulates around the insertion site.^{10,11}

The dressing must be replaced 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.

Gauze dressings must be replaced at least every two days.

Clear transparent dressings must be replaced at least every seven days.¹¹

Line sites must be monitored daily for any pain, redness or purulence that may be suggestive of local infection.

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.



7.4 The use of catheters and dressings that have associated antimicrobial activity

Recommendations

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.¹²

Antimicrobial-impregnated catheters or those with antimicrobial properties may be considered in environments in which the rate of CLABSI is not decreasing, and lines are likely to remain in place for more than five days.¹³

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.

7.5 The management of lines and administration sets

Recommendations

Any fat-containing emulsion (nutritional lipid emulsions or drugs with a fat emulsion-based vehicle) infusion sets in continuous use do not need to be changed more often than four days, but at least every seven days^{14,15} for patients not receiving blood or blood products.

Infusion sets that have contained blood or blood products or fat-containing emulsions must be changed within 24 hours of initiating the infusion.¹⁶

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.¹⁷

Needleless connectors and access ports on administration sets must be cleaned with 70% alcohol, tincture of iodine or chlorhexidine, prior to injection or connection.¹⁸

Rationale

The infusion of blood or blood products or fat-containing emulsions increases the risk of infection.

Unless strict cleaning procedures are adhered to, the use of needleless connectors are no safer from an infection perspective.

7.6 The insertion of peripheral catheters

Recommendations

Strict hand hygiene must be observed before and after the placement, removal or palpation of the catheter-insertion site.^{19,20}

Strict hand hygiene is also required before and after accessing or dressing a catheter.^{19,20}

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.²¹

Sterile gloves should be used for the insertion of arterial and umbilical catheters.⁵

Clean gloves must be worn when changing the dressings on intravascular catheters.²²

Skin preparation with 70% alcohol, tincture of iodine or chlorhexidine is acceptable for peripheral venous catheter insertion.^{8,9}

A cleaning solution that contains more than 0.5% chlorhexidine in alcohol should be used when inserting arterial catheters.^{8,9}

The upper extremity should be used for the insertion of venous catheters in adults.²³

The upper or lower extremities, or the scalp, may be used as a catheter insertion site in paediatric patients.²³

The radial or *dorsalis pedis* site is preferred over the femoral or axillary site to reduce the risk of infection in adults.²⁴

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.²⁵

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.^{26,27}

References

1. Pronovost PJ, Needham D, Berenholtz S, et al. An intervention to decrease catheter-related bloodstream infections in the ICU. *N Engl J Med*. 2006;355(26):2725-2732.
2. Merrer J, De Jonghe B, Golliot F, et al. Complications of femoral and subclavian venous catheterization in critically ill patients: a randomized controlled trial. *JAMA*. 2001;286(6):700-707.
3. Hind D, Calvert N, McWilliams R, et al. Ultrasonic locating devices for central venous cannulation: meta-analysis. *BMJ*. 2003;327(7411):361.
4. Yeung C, May J, Hughes R. Infection rate for single lumen v triple lumen subclavian catheters. *Infect Control Hosp Epidemiol*. 1988;9(4):154-158.
5. Raad II, Hohn DC, Gilbreath BJ, et al. Prevention of central venous catheter-related infections by using maximal sterile barrier precautions during insertion. *Infect Control Hosp Epidemiol*. 1994;15(4 Pt 1):231-238.
6. Sherertz RJ, Ely EW, Westbrook DM, et al. Education of physicians in-training can decrease the risk for vascular catheter infection. *Ann Intern Med*. 2000;132(8):641-648.
7. Carrer S, Bocchi A, Bortolotti M, et al. Effect of different sterile barrier precautions and central venous catheter dressing on the skin colonization around the insertion site. *Minerva Anestesiol*. 2005;71(5):197-206.



8. Maki DG, Ringer M, Alvarado CJ. Prospective randomised trial of povidone-iodine, alcohol and chlorhexidine for prevention of infection associated with central venous and arterial catheters. *Lancet*. 1991;338(8763):339-343.
9. Mimos O, Pieroni L, Lawrence C, et al. Prospective, randomized trial of two antiseptic solutions for prevention of central venous or arterial catheter colonization and infection in intensive care unit patients. *Crit Care Med*. 1996;24(11):1818-1823.
10. Maki DG, Stolz SS, Wheeler S, Mermel LA. A prospective, randomized trial of gauze and two polyurethane dressings for site care of pulmonary artery catheters: implications for catheter management. *Crit Care Med*. 1994;22(11):1729-1737.
11. Madeo M, Martin CR, Turner C, et al. A randomized trial comparing Arglaes (a transparent dressing containing silver ions) to Tegaderm (a transparent polyurethane dressing) for dressing peripheral arterial catheters and central vascular catheters. *Intensive Crit Care Nurs*. 1998;14(4):187-191.
12. Ho KM, Litton E. Use of chlorhexidine-impregnated dressing to prevent vascular and epidural catheter colonization and infection: a meta-analysis. *J Antimicrob Chemother*. 2006;58(2):281-287.
13. Brun-Buisson C, Doyon F, Sollet JP, et al. Prevention of intravascular catheter-related infection with newer chlorhexidine-silver sulfadiazine-coated catheters: a randomized controlled trial. *Intensive Care Med*. 2004;30(5):837-843.
14. Gillies D, Wallen MM, Morrison AL, et al. Optimal timing for intravenous administration set replacement. [Cochrane review]. In: *The Cochrane Library*, Issue 4 2005. Oxford: Update Software.
15. Maki DG, Botticelli JT, LeRoy ML, Thielke TS. Prospective study of replacing administration sets for intravenous therapy at 48- vs 72-hour intervals. 72 hours is safe and cost-effective. *JAMA*. 1987;258(13):1777-1781.
16. Maki DG, Martin WT. Nationwide epidemic of septicemia caused by contaminated infusion products. IV. Growth of microbial pathogens in fluids for intravenous infusions. *J Infect Dis*. 1975;131(3):267-272.
17. Bennett SN, McNeil MM, Bland LA, et al. Postoperative infections traced to contamination of an intravenous anesthetic, propofol. *N Engl J Med*. 1995;333(3):147-154.
18. Casey AL, Burnell S, Whinn H, et al. A prospective clinical trial to evaluate the microbial barrier of a needleless connector. *J Hosp Infect*. 2007;65(3):212-218.
19. Coopersmith CM, Rebmann TL, Zack JE, et al. Effect of an education program on decreasing catheter-related bloodstream infections in the surgical intensive care unit. *Crit Care Med*. 2002;30(1):59-64.
20. Boyce JM, Pittet D. Guideline for hand hygiene in health-care settings: recommendations of the Healthcare Infection Control Practices Advisory Committee and the HICPAC/SHEA/APIC/IDSA Hand Hygiene Task Force. *Infect Control Hosp Epidemiol*. 2002;23(12 Suppl):S3-S40.
21. Pittet D, Dharan S, Touveneau S, et al. Bacterial contamination of the hands of hospital staff during routine patient care. *Arch Intern Med*. 1999;159(8):821-826.
22. Abi-Said D, Raad I, Umphrey J, et al. Infusion therapy team and dressing changes of central venous catheters. *Infect Control Hosp Epidemiol*. 1999;20(2):101-105.
23. Maki DG, Goldman DA, Rhame FS. Infection control in intravenous therapy. *Ann Intern Med*. 1973;79(6):867-887.
24. Traore O, Liotier J, Souweine B. Prospective study of arterial and central venous catheter colonization and of arterial and central venous catheter-related bacteremia in intensive care units. *Crit Care Med*. 2005;33(6):1276-1280.
25. Maki DG, Ringer M. Risk factors for infusion-related phlebitis with small peripheral venous catheters. A randomized controlled trial. *Ann Intern Med*. 1991;114(10):845-854.
26. 2011 guidelines for the prevention of intravascular catheter-related infections. Centers for Disease Control and Prevention. CDC [homepage on the Internet]. Available from: <http://www.cdc.gov/hicpac/BSI/BSI-guidelines-2011.html>
27. Guideline for disinfection and sterilisation in healthcare facilities, 2008. Healthcare Infection Control Practices Advisory Committee. HICPAC [homepage on the Internet]. Available from: http://www.cdc.gov/hicpac/Disinfection_Sterilization/acknowledg.html



8. Infection control recommendations for regional anaesthesia

8.1 Spinal, epidural and caudal procedures

A historical and physical examination and review of the relevant laboratory studies should be conducted to identify patients who may be at risk of infectious complications. 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 bacteremia, prophylactic pre-procedural antibiotic therapy should be considered.

Aseptic techniques 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), 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 chlorhexidine with alcohol, should be used for skin preparation, and adequate time allowed for drying.
- 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 is 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 1 in 145 000.¹ Although rare, the consequences of these complications are disastrous. In the latest *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.²

The recommendations are based largely on a practice advisory produced by the American Society of Anesthesiologists (ASA) in 2010.³ Guidelines produced by the Association of Anaesthetists

of Great Britain and Ireland (AAGBI),⁴ and Australian and New Zealand College of Anaesthetists (ANZA) make similar recommendations.⁵

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.³

In the discussion which follows, any point about which specific commentary has not been made has been included because of expert opinion.

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.³

Jewellery

Watches and rings should be removed during hand washing.^{6,7} Higher microbial counts have been found in healthcare workers who do not remove rings after hand washing.⁷

Gowns

ASA members and consultants were equivocal with regard to the wearing of gowns.³ The AAGBI includes gowns as part of its maximal barrier precautions, which it recommends for neuraxial techniques.⁴ The ANZA guidelines also suggest that members wear gowns while performing neuraxial anaesthesia.⁵ There are insufficient data to make definitive recommendations with regard 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.⁸ 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.⁹ 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.¹⁰ 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.¹¹ Therefore, in line with a recent CDC recommendation, the use of a surgical face mask is recommended when performing a neuraxial procedure.¹²

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.^{13,14}

The antimicrobial action of alcohol is denaturation of proteins and its dehydrating properties. Therefore, it must be allowed to dry completely before the procedure.¹⁵

Bacterial filter

Three observational studies showed that infections and epidural abscesses could occur in the presence of micropore filters.¹⁶⁻¹⁸ 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.¹⁹⁻²⁷ The literature has not identified a specific duration associated with increased infection, so the recommendation is that catheters should not remain in situ longer than is clinically necessary.³

References

- Ruppen W, Derry S, McQuay H, et al. Incidence of epidural hematoma, infection, and neurologic injury in obstetric patients with epidural anesthesia/ analgesia: meta-analysis. *Anesthesiology*. 2006;105(2):394-399.
- Saving mothers 2008-2010: Fifth report on the Confidential Enquiries Into Maternal Deaths in South Africa. Pretoria: Department of Health; 2012.
- American Society of Anesthesiologists Task Force. Practice advisory for the prevention, diagnosis and management of infectious complications associated with neuraxial techniques. *Anesthesiology*. 2010;112(3):530-545.
- Association of Anaesthetists of Great Britain and Ireland. AAGBI safety guideline 2008: infection control in anaesthesia. AAGBI [homepage on the Internet]. Available from: http://www.aagbi.org/sites/default/files/infection_control_08.pdf Australian and New Zealand College of Anaesthetists. Guidelines on infection control in anaesthesia. ANZCA [homepage on the Internet]. 2005. Available from: <http://www.anzca.edu.au/resources/professional-documents/pdfs/ps28-2013-guidelines-on-infection-control-in-anaesthesia.pdf>
- Hartley JC, Mackay AD, Scott GM. Wrist watches must be removed before washing hands. *Br Med J* 1999;318(7179):328.
- Salisbury DM, Hutfilz P, Treen LM, et al. The effect of rings on microbial load of health care workers' hands. *Am J Infect Control*. 1997;25(1):24-27.
- Baer ET. Post-dural puncture bacterial meningitis. *Anesthesiology*. 2006;105(2):381-393.
- Schneeberger PM, Janssen M, Voss A. Alpha-hemolytic streptococci: a major pathogen of iatrogenic meningitis following lumbar puncture. Case reports and a review of the literature. *Infection*. 1996;24(1):29-33.
- North JB, Brophy BP. Epidural abscess: a hazard of spinal epidural anaesthesia. *Aust N Z J Surg*. 1979;49(4):484-485.
- Phillips BJ, Fergusson S, Armstrong P, et al. Surgical face masks are effective in reducing bacterial contamination caused by dispersal from the upper airway. *Br J Anaesth*. 1992;69(4):407-408.
- CDC clinical reminder: spinal injection procedures performed without a facemask

- Centers for Disease Control and Prevention; 2013.
- Kinirons B, Mimoz O, Lafendi L, et al. Chlorhexidine versus povidone iodine in preventing colonization of continuous epidural catheters in children: a randomized, controlled trial. *Anesthesiology*. 2001;94(2):239-244.
- Shibata S, Shibata I, Tstudy A, et al. Comparative effects of disinfectants on the epidural needle/catheter contamination with indigenous skin bacterial flora. *Anesthesiology*. 2004;101:A1363.
- Guideline for disinfection and sterilization in healthcare facilities. Centers for Disease Control and Prevention [homepage on the Internet]. 2008. Available from: http://www.cdc.gov/hicpac/Disinfection_Sterilization/toc.html
- Bevacqua BK, Slucky AV, Cleary WF. Is postoperative intrathecal catheter use associated with central nervous system infection? *Anesthesiology*. 1994;80(6):1234-1240.
- Seth N, Macqueen S, Howard RF. Clinical signs of infection during continuous postoperative epidural analgesia in children. The value of catheter tip culture. *Paediatr Anaesth*. 2004;14(12):996-1000.
- James FM, George RH, Raiem H, White GJ. Bacteriologic aspects of epidural analgesia. *Anesth Analg*. 1976;55(2):187-190.
- Erdine S, Aldemir T. Long-term results of peridural morphine in 225 patients. *Pain*. 1991;45(2):155-159.
- Sillevis Smitt P, Tsafka A, Teng-van de Zande F, et al. Outcome and complications of epidural analgesia in patients with chronic cancer pain. *Cancer*. 1998;83(9):2015-2022.
- James FM, George RH, Raiem H, White GJ. Bacteriologic aspects of epidural analgesia. *Anesth Analg*. 1976;55(2):187-190.
- De Jong, Kansen PJ. A comparison of epidural catheters with or without subcutaneous injections ports for treatment of cancer pain. *Anesth Analg*. 1994;78(1):94-100.
- De Leon-Casasola OA, Parker B, Lema M, et al. Postoperative epidural bupivacaine-morphine therapy: experience with 4227 surgical cancer patients. *Anesthesiology*. 1994;81(2):368-375.
- Holt HM, Andersen SS, Andersen O, et al. Infections following epidural catheterization. *J Hosp Infect*. 1995;30(4):253-260.
- Mann TJ, Orlikowski CE, Gurrin LC, Keil AD. The effect of the biopatch, a chlorhexidine impregnated dressing, on bacterial colonization of epidural catheter exit sites. *Anaesth Intensive Care*. 2001;29(6):600-603.
- Mishra S, Bhatnagar S, Srikanti M, Gupta D. Clinical implication of routine bacterial culture from epidural catheter tips in postoperative cancer patients: A prospective study. *Anaesthesia*. 2006;61(9):878-882.
- Wang LP, Hauerberg J, Schmidt JF. Incidence of spinal epidural abscess after epidural analgesia: a national 1-year survey. *Anesthesiology*. 1999;91(6):1928-1936.

8.2 Peripheral nerve blocks

Maximal barrier precautions are generally not necessary. Maximum barrier precautions should be used only 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.

The insertion of a perineural catheter

Use a sterile occlusive dressing.

Limit disconnection and reconnection of the delivery system.

Catheters 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, e.g. the ultrasound machine and probe should be wiped with a single-use towel to remove visible soiling, and then wiped with another single-use towel that has been soaked in an appropriate disinfectant, e.g. 70% isopropyl alcohol, then allowed to dry.

Product information should be consulted with regard to which cleaning agents are appropriate for the specific machine or probe.

Use single-use, sterile gel, e.g. a 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 infection of 0-3.2% and abscess formation of 0-0.9%).¹ The risk is smaller still for "single-shot" blocks. There have been only occasional case reports in the literature.²

Unless a perineural catheter is to be placed, or the patient is immunocompromised, both the AAGBI and ANZA guidelines agree that maximal barrier precautions are unnecessary when performing peripheral nerve blocks.^{3,4}

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.⁵⁻⁸ 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.

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).⁹ Ultrasound-guided regional techniques involves 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 Spaulding's system, requires full sterilisation or the use of a sterile sheath, followed by prolonged immersion with HLD.⁹

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.¹⁰

Recently, Chuan et al assessed the effectiveness of a simple three-step decontamination protocol using 70% isopropyl alcohol as the disinfectant for ultrasound equipment used to perform single-shot, peripheral ultrasound-guided nerve blocks. They concluded that acceptable disinfection was consistently achieved in all of the cases.¹⁰

The same author suggests that a sterile transducer sheath is used, in addition to what has been described, for the following:

- Immunocompromised patients.
- Patients with multi-resistant organisms.
- Patients undergoing perineural catheter placement.
- When there is a risk of blood contamination of the probe.¹¹

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.

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.¹² In 2012, Oleszkowicz et al proposed similar guidelines for the USA.¹³ In line with these, the use of sterile, single-use, ultrasound gel for invasive procedures is recommended.

References

1. Capdevila X, Bringuier S, Borgeat A. Infectious risk of continuous peripheral nerve blocks. *Anesthesiology*. 2009;110(1):182-188.
2. Nseir S, Pronnier P, Soubrier S, et al. Fatal streptococcal necrotizing fasciitis as a complication of axillary brachial plexus block. *Br J Anaesth*. 2004;92(3):427-429.
3. Association of Anaesthetists of Great Britain and Ireland. AAGBI safety guideline 2008: Infection control in anaesthesia. AAGBI [homepage on the Internet]. Available from: http://www.aagbi.org/sites/default/files/infection_control_08.pdf
4. Australian and New Zealand College of Anaesthetists. Guidelines on infection control in anaesthesia. ANZCA [homepage on the Internet]. 2005. Available from: <http://www.anzca.edu.au/resources/professional-documents/pdfs/ps28-2013-guidelines-on-infection-control-in-anaesthesia.pdf>
5. Muradali D, Gold WL, Phillips A, Wilson S. Can ultrasound probes and coupling gel be a source of nosocomial infection in patients undergoing sonography? An in vivo and in vitro study. *AJR Am J Roentgenol*. 1995;164(6):1521-1524.
6. Ohara T, Itoh Y, Itoh K. Ultrasound instruments as possible vectors of staphylococcal infection. *J Hosp Infect*. 1998;40(1):73-77.
7. Gaillot O, Maruejols C, Abachin E, et al. Nosocomial outbreak of *Klebsiella pneumoniae* producing SHV-5 extended-spectrum beta-lactamase, originating from a contaminated ultrasonography coupling gel. *J Clin Microbiol*. 1998;36(5):1357-1360.
8. Weist K, Wendt C, Petersen LR, Versmold H, Ruden H. An outbreak of pyoderma among neonates caused by ultrasound gel contaminated with methicillin-susceptible *Staphylococcus aureus*. *Infect Control Hosp Epidemiol*. 2000;21(12):761-764.
9. Rutala W, Weber D. Guideline for disinfection and sterilization in healthcare facilities. Department of Health and Human Services USA; 2008.
10. Chuan A, Tiong C, Maley M, et al. Decontamination of ultrasound equipment used for peripheral ultrasound-guided regional anaesthesia. *Anaesth Intensive Care*. 2013;41(4):529-534.
11. Aseptic technique. Ultrasound block.com [homepage on the Internet]. Available from: <http://www.ultrasoundblock.com/index.php/nerve-block-controversies/nerve-block-aseptic-technique>
12. Government of Canada. Serious risk of infection from ultrasound and medical gels: revision. 2004 [homepage on the Internet]. Available at: <http://www.canadiensante.gc.ca/recall-alert-rappel-avis/hc-sc/2004/14289a-eng.php>.
13. Oleszkowicz SC, Chittick P, Rultrasoundso V, et al. Infections associated with use of ultrasound transmission gel: proposed guidelines to minimize risk. *Infect Control Hosp Epidemiol*. 2012;33(12):1235-1237.

9. Prevention of surgical site infection for the anaesthetist

9.1 Antibiotic prophylaxis for surgical procedures

Antibiotic prophylaxis should be administered before:

- “Clean” surgery involving the placement of prosthesis or implants.
- “Clean-contaminated” surgery.
- “Contaminated” surgery.¹

Do not give antibiotic prophylaxis for clean, non-prosthetic uncomplicated surgery.^{1,2}

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.³

Use your local antibiotic formulary, and always consider potential adverse effects when giving antibiotics for prophylaxis.¹

Give a single dose of antibiotic prophylaxis intravenously on starting anaesthesia 30 minutes before skin incision, but not more than one hour before it. However, give antibiotic prophylaxis earlier for operations in which a tourniquet is used.^{1,2}

A second dose of an antibiotic with a relatively short half-life, e.g. cephazolin, is often recommended for prolonged procedures.^{1,2}

Administer the initial dose of the prophylactic antimicrobial agent by the intravenous route, timed such that a bactericidal concentration of the drug is present in the serum and tissue at the time of incision. Maintain therapeutic levels of the drug in serum and tissue throughout the operation.

Consider antibiotic treatment in patients who have undergone surgery on a dirty or infected wound.¹

Rationale

Antibiotic prophylaxis reduces bacterial inoculum at the time of surgery. It significantly decreases the rate of bacterial contamination of the surgical site.²

Surgical wound classification is as follows:^{1,2}

- “Clean” surgery: “Clean” surgery is when there is no break in aseptic technique and the respiratory, gastrointestinal or genitourinary tracts are not breached.
- “Clean-contaminated” surgery: This extends to the oropharynx, sterile genitourinary or biliary tract, the gastrointestinal or respiratory tracts, or if there has been a minor breach in the aseptic technique.
- “Contaminated” surgery: The presence of acute inflammation, infected biliary secretions, infected urine or gross contamination from the gastrointestinal tract.
- “Dirty” surgery: If an established infection exists, and therapeutic antibiotics are administered based on the susceptibility of bacterial isolates grown from culture.

Antibiotics have associated risks. Adverse effects include gastrointestinal symptoms (nausea, vomiting or diarrhoea) 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.²

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.⁴ Surgeries that involve the bowel necessitate Gram-negative and anaerobic coverage, for which ceftiofexim is appropriate. Vancomycin may be the prophylaxis of choice when a “cluster of methicillin-resistant *S. aureus* mediastinitis, or incisional surgical site infection owing to methicillin-resistant, coagulase-negative staphylococci, has been detected.⁴

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.²

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.²

The WHO initiative, “Safe surgery saves lives” surgical safety checklist emphasises the inclusion of antibiotic prophylaxis given 60 minutes before skin incision.⁵

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.¹

Prolonged antibiotic prophylaxis, extending after the surgical procedure, has not been shown to be effective.

Early trials of cephalosporins revealed cross-reactivity in penicillin-allergic patients where there was an anaphylaxis rate of approximately 8%. The standard of care became the avoidance of cephalosporins in these patients.

Multiple studies have shown the relative clinical safety of administering cephalosporins to penicillin-allergic patients.⁴

If the potential for allergic reaction to a cephalosporin is deemed to be high, e.g. previous collapse or severe anaphylaxis to penicillin, clindamycin or vancomycin may be used.⁴



Inform patients before the operation if they are going to receive antibiotic prophylaxis, and afterwards if they have been given antibiotics prophylaxis during their operation.¹

References

1. National Institute for Health and Clinical Excellence. Surgical site infection prevention and treatment of surgical site infection. NICE; 2008.
2. Gifford C, Christelis N, Cheng A. Preventing postoperative infection: the anaesthetist's role. *Contin Educ Anaesth Crit Care Pain*. 2011;11(5):151-156.
3. Mangram AJ, Horan TC, Pearson ML, et al. Guideline for prevention of surgical site infection. *Infect Control Hosp Epidemiol*. 1999;20(4):250-278.
4. Mauerman WJ, Nemergut EC. The anesthesiologist's role in the prevention of surgical site infections. *Anesthesiology*. 2006;105(2):413-421.
5. World Health Organization. Safe surgery saves lives surgical safety checklist. WHO [homepage on the Internet]. Available from: www.who.int/patientsafety/safesurgery/.

9.2 Physiological parameter homeostasis to reduce the risk of surgical site infections

Recommendations

Maintain normoxia throughout the perioperative period.

Give supplemental oxygen to maintain arterial saturation above 95%.

Optimise the components that relate to oxygen delivery, i.e. the haemoglobin levels, fractional inspiration of oxygen and cardiac output.

Maintain optimal perfusion to the operative site.

Avoid hypothermia.

Avoid severe hyperglycaemia.

Minimise blood transfusion and use leucodepleted blood in high-risk patients.

Rationale

Oxygenation and perfusion

Oxygen is a very important component in the oxidative killing of microorganisms. It has been shown that phagocytosing neutrophils have a burst of oxygen consumption that is caused by the nicotinamide adenine dinucleotide phosphate-oxidase complex. This leads to the formation of superoxide and other reactive oxygen species that are responsible for destruction of microbes.¹

Oxygen is also a very important component of wound healing.² Adequate wound healing is necessary to protect the underlying tissue from direct exposure to microbes, with the resultant risk of surgical site infection. It had been shown that supplemental oxygen accelerates the growth of new blood vessels into the damaged area.³ It also helps to increase levels of vascular endothelial growth factor⁴ and promotes contraction of the wound by facilitating the differentiation of fibroblasts into myofibroblasts.⁵

A fresh trauma or surgical wound is an acutely hypoxic environment because of the disruption of blood vessels. The centre of a wound can have a PaO₂ as low as 0-10 mmHg, compared a PaO₂ of approximately 60 mmHg around the edges.^{2,6}

It is necessary to ensure that the components of the oxygen delivery (DO₂) equation are optimised to maintain optimal oxygen delivery to the damaged tissue:⁷

$$DO_2 = \text{cardiac output} \times \text{oxygen content in blood (CaO}_2\text{)}$$

$$CaO_2 = (\text{haemoglobin} \times \text{sats} \times 0.34) + (\text{PaO}_2 \times 0.003).$$

A number of studies have examined the influence of hyperoxia on the rate of surgical site infections. The results have been mixed, either favouring higher oxygen concentrations,⁸⁻¹⁴ showing no statistical significant difference,¹⁵⁻¹⁸ or even demonstrating that high oxygen concentrations increase the risk of surgical site infections.¹⁹ The majority of studies were carried out on patients presenting for colorectal surgery. Most of these studies showed a benefit. The results were more difficult to interpret in other surgical fields. There was no difference in obstetric patients presenting for Caesarean section, between those who had received more than 60% oxygen and those who had received less than 40%.¹⁶

Hyperglycaemia

Insulin-resistant hyperglycaemia is part of the metabolic stress response to trauma and surgery. This stress-induced hyperglycaemia is owing to increased sympathomimetic activity and the release of counterregulatory hormones and proinflammatory cells.

The effect of hyperglycaemia on the immune system is that it impairs host defences, decreases polymorphonuclear leukocyte mobilisation, decreases chemotaxis and leads to decreased phagocytic activity. Subsequently, the release of superoxide and radical oxygen species is compromised, with decreased oxidative killing.

A number of studies²⁰⁻³³ have shown a decreased incidence of infection when glucose was controlled. The controversial issue is finding the absolute value to target for blood glucose concentration. May³⁴ advocates a blood glucose level of less than 200 mg/dl (13 mmol/l), while others like Butler et al³⁵ target tight control glucose levels between 80 mg/dl (4.4 mmol/l) and 110 mg/dl (6.2 mmol/l).

The famous Van den Berghe trials^{30,36} showed benefit with intensive insulin therapy which targeted blood glucose levels between 80 and 110 mg/dl (4.4-6.1 mmol/l). The benefit was more pronounced in the setting of the surgical ICU. However, the Normoglycemia in Intensive Care Evaluation and Surviving Using Glucose Algorithm Regulation (NICESUGAR) trial³⁷ demonstrated that there was increased mortality associated with intensive insulin therapy, and most notably an increase in the incidence of hypoglycaemic events. The authors suggest a blood glucose target of less than 180 mg/dl (10 mmol/l). It has to



be noted that most of the studies examined critically ill patients. The effect of hyperglycaemia on healthy patients hasn't been studied extensively.

Hypothermia

Hypothermia is defined as a core body temperature of less than 36°C. Hypothermia commonly occurs during anaesthesia due to several factors, such as anaesthetic-induced peripheral vasodilatation, exposure of the body during surgical preparation and the exposure of body cavities to ambient temperature.³⁸

Hypothermia had been implicated in the pathogenesis of surgical site infection. Firstly, it causes vasoconstriction, and therefore has a negative impact on the delivery of oxygen to the site of injury. As discussed previously, oxygen is very important in the oxidative killing of microbes. Secondly, it has a direct impact on the immune system. Clardy et al³⁹ showed that when exposed to hypothermic conditions, neutrophils in vitro had a decreased ability to migrate towards the chemotactic stimulus, had a decreased ability to ingest *Staphylococci*, and also failed to be metabolically activated by superoxide production. There was also a significant correlation between the T-lymphocyte-dependent production of immunoglobulins and temperature.⁴⁰ Thirdly, hypothermia also impairs coagulation. The increased risk of bleeding can either lead to postoperative haematoma that may become infected, or to an increased need for blood transfusion. Blood transfusion also leads to depression of the immune system.

Various studies showed a decrease in surgical site infections when normothermia was maintained.⁴¹⁻⁴³

Flores-Maldonado et al showed a reduction of surgical site infections from 11.5% in the hypothermic group to 2% in the normothermic group. Seamon et al also showed that an intraoperative temperature below 35°C adversely affected the surgical site infection rate.

In 1999, Barone et al⁴⁴ questioned an earlier study by Kurz et al, carried out on colorectal surgery patients. They questioned the methodology of the study, and subsequently retrospectively reviewed colorectal surgery patients' records and found no difference between the two groups.

Blood transfusion-related immunomodulation

Allogenic blood transfusion has been associated with an increased risk of postoperative bacterial infection by as much as 200-1 000%.⁴⁵ However, it is difficult to establish whether or not this association was causal as other factors such as anaesthesia, shock and hypothermia could have potentially played a role too. When evaluating animal models, Waymack et al⁴⁶ found that blood transfusion led to a decrease in macrophage migration, and to an increase in the production of the immunosuppressive prostaglandin E. Various other immunomodulatory effects have also been demonstrated,⁴⁷ such as decreased interleukin 2 secretion, decreased natural killer cell activity,⁴⁸ decreased

delayed type hypersensitivity reactions, decreased CD₄ and CD₈ ratios and decreased macrophage functions.

Jensen et al⁴⁸ conducted a study on elective colorectal surgery patients and compared the incidence of surgical site infections of patients receiving whole blood, those receiving leucodepleted blood, and those who didn't require blood transfusion. He found an incidence of 23% in patients receiving whole blood versus an incidence of 2% in both the non-transfusion group and the group that received leuco-depleted blood.

References

- Hampton MB, Kettle AJ, Winterbourn CC. Inside the neutrophil phagosome: oxidants, myeloperoxidase and bacterial killing. *Blood*. 1998;92(9):3007-3017.
- Ansgar AO, Barie PS, Faist E, et al. Current issues in the prevention and management of surgical site infection: part 1. *Surg Infect (Larchmt)*. 2002;3 Suppl 1:S1-57.
- Knighton DR, Silver IA, Hunt TK. Regulation of wound healing angiogenesis-effect of oxygen gradients and inspired concentrations. *Surgery*. 1981;90(2):262-270.
- Patel V, Chivukala I, Roy S, et al. Oxygen: from the benefits of inducing VEGF expression to managing the risk of hyperbaric stress. *Antioxid Redox Signal*. 2005;7(9-10):1377-1387.
- Rodriguez PG, Felix FN, Woodley DT, Shim ER. The role of oxygen in wound healing: a review of the literature. *Dermatol Surg*. 2008;34(9):1159-1169.
- Gordillo GM, Sen CK. Revisiting the essential role of oxygen in wound healing. *Am J Surg*. 2003;186(3):259-263.
- McLellan SA, Walsh TS. Oxygen delivery and hemoglobin. *Contin Educ Anaesth Crit Care Pain*. 2004;4(4):123-126.
- Greif R, Akça Q, Hoin EP, et al. Supplemental peri-operative oxygen to reduce the incidence of surgical wound infection. *N Eng J Med*. 2000;342(3):161-167.
- Belda FJ, Aguilera L, García de la Asunción J, et al. Supplemental peri-operative oxygen and the risk of surgical wound infections: a randomized controlled trial. *JAMA*. 2005;294(16):2035-2042.
- Havaguimian F, Lysakowski C, Ella N, Tramèr MR. Effect of intra-operative high fraction of inspired oxygen on surgical site infections, post operative nausea and vomiting and pulmonary function: a systematic review and meta analysis of randomized controlled trials. *Anesthesiology*. 2013;119(2):303-316.
- Al-Niami A, Safdar N. Supplemental peri operative oxygen for reducing surgical site infections: a meta analysis. *J Eval Clin Pract*. 2009;15(2):360-365.
- Chura JC, Boyd A, Argenta PA. Surgical site infection and supplemental peri-operative oxygen in colorectal surgery patients: a systematic review. *Surg Infect (Larchmt)*. 2007;8(4):455-461.
- Qadan M, Akça O, Mahid SS, et al. Perioperative supplemental oxygen therapy and surgical site infections: a meta-analysis of randomized controlled trials. *Arch Surg*. 2009;144(4):359-366.
- Kao LS, Millas SG, Pedraza C, et al. Should peri-operative supplemental oxygen be routinely recommended for surgery patients? A Bayesian meta-analysis. *Ann Surg*. 2012;256(6):894-901.
- Tagioka B, Galvagno S, Sumida S, et al. The role of peri-operative high inspired oxygen therapy in reducing surgical site infections: a meta-analysis. *Anesth Analg*. 2012;114(2):334-342.
- Klingel ML, Patel SV. A meta-analysis of the effect of inspired oxygen concentration on the incidence of surgical site infection following cesarean section. *Int J Obstet Anesth*. 2013; 22(2):104-112.
- Patel SV, Coughlin SC, Malthaner RA. High concentration oxygen and surgical site infection in abdominal surgery: a meta-analysis. *Can J Surg*. 2013;56(4):E82-E90.
- Brar MS, Brar SS, Dixon E. Peri-operative supplemental oxygen in colorectal patients: a meta-analysis. *J Surg Res*. 2011;166(2):227-235.
- Pryor KO, Fahey TJ 3rd, Lien CA, Goldstein PA. Surgical site infection and the routine use of peri-operative hyperoxia in a general surgical population: a randomized control trial. *JAMA*. 2004;291(1):79-87.
- Fietsam R, Bassett J, Glover JL. Complications of coronary artery surgery in diabetic patients. *Am Surg*. 1991;57(9):551-557.



21. Zerr KJ, Furnary AP, Grunkemeier GL, et al. Glucose control lowers the risk of wound infection in diabetics after open-heart operations. *Ann Thorac Surg.* 1997;63(2):356-61.
22. Pomposelli JJ, Baxter JK, Babineau TJ, et al. Early postoperative glucose control predicts nosocomial infection rate in diabetic patients. *J Parenter Enteral Nutr.* 1998;22(2):77-81.
23. Golden SH, Peart-Vigilance C, Kao WHL, Brancati FL. Perioperative glycemic control and the risk of infectious complications in a cohort of adults with diabetes. *Diabetes Care.* 1999;22(9):1408-1414.
24. Furnary AP, Zerr KJ, Grunkemeier GL, Starr A. Continuous intravenous insulin infusion reduces the incidence of deep sternal wound infection in diabetic patients after cardiac surgical procedures. *Ann Thorac Surg.* 1999;67:352-362.
25. Trick WE, Scheckler WE, Tokars JE, et al. Modifiable risk factors associated with deep sternal site infection after coronary artery bypass grafting. *J Thorac Cardiovasc Surg.* 2000;119(1):108-114.
26. Trick WE, Scheckler WE, Tokars JE, et al. Risk factors for radial artery harvest site infection following coronary artery bypass graft surgery. *Clin Infect Dis.* 2000;30(2):270-275.
27. Latham R, Lancaster AD, Covington JF, et al. The association of diabetes and glucose control with surgical-site infections among cardiothoracic surgery patients. *Infect Control Hosp Epidemiol.* 2001;22(10):607-612.
28. M, Pasaoglu I, Demircin M, Oc M. Perioperative hyperglycemia is a strong correlate of postoperative infection in type II diabetic patients after coronary artery bypass grafting. *Endocr J.* 2002;49(5):531-537.
29. Estrada CA, Young JA, Nifong LW, Chitwood WR. Outcomes and perioperative hyperglycemia in patients with or without diabetes mellitus undergoing coronary artery bypass grafting. *Ann Thorac Surg.* 2003;75(5):1932-1939.
30. Van den Berghe G, Wouters P, Weekers F, et al. Intensive insulin therapy in critically ill patients. *N Engl J Med.* 2001;345(19):1359-1367.
31. Krinsley JS. Association between hyperglycemia and increased hospital mortality in a heterogeneous population of critically ill patients. *Mayo Clin Proc.* 2003;78(12):1471-1478.
32. Finney SJ, Zekveld C, Elia A, Evans TW. Glucose control and mortality in critically ill patients. *JAMA.* 2003;290(15):2041-2047.
33. Krinsley JS. Effect of an intensive glucose management protocol on the mortality of critically ill adult patients. *Mayo Clin Proc.* 2004;79(8):992-1000.
34. AK, Kauffmann RM, Collier BR. The place for glycaemic control in the surgical patient. *Surg Infect.* 2011; 12(5):405-418.
35. Butler SO, Btaiche IF, Alaniz C. Relationship between hyperglycaemia and infection in critically ill patients. *Pharmacotherapy.* 2005;25(7):963-976.
36. Van den Berghe G, Wilmer A, Hermans G, et al. Intensive insulin therapy in the medical ICU. *N Engl J Med.* 2006;354(5):449-461.
37. Finfer S, Chittock DR, et al. NICE-SUGAR study investigators. Intensive versus conventional glucose control in critically ill patients. *N Engl J Med.* 2009;360(13):1283-1297.
38. Perioperative hypothermia (inadvertent) (CG65). National Institute for Health Care and Excellence. NICE [homepage on the Internet]. Available from: www.nice.org.uk/CG65
39. Clardy CW, Edwards KM, Gay JC. Increased susceptibility to infection in hypothermic children: possible role of acquired neutrophil dysfunction. *Pediatr Infect Dis.* 1985;4(4):379-382.
40. Horosz B, Malec-Milewska M. Inadvertent intraoperative hypothermia. *Anaesthesiology Intensive Ther.* 2013;45(1):38-43.
41. Kurz A, Sessler DI, Lenhardt R. Perioperative normothermia to reduce the incidence of surgical wound infections and shorten hospitalization. Study of Wound Infection and Temperature Group. *N Engl J Med.* 1996;334(19):1209-1215.
42. Flores Maldonado A, Medina-Escobedo CE, Ríos-Rodríguez HM, Fernández-Domínguez. Mild intraoperative hypothermia and the risk of wound infection. *Arch Med Res.* 2001;32(3):227-231.
43. Seamon MJ, Wobb J, Gaughan JP, et al. The effects of intraoperative hypothermia on surgical site infection: an analysis of 524 trauma laparotomies. *Ann Surg.* 2012;255(4):789-795.
44. Barone JE, Tucker JB, Cecere J, Yoon MJ, et al. Hypothermia does not result in more complications after colon surgery. *Am Surg.* 1999;65(4):356-359.
45. Blumberg N, Heal JM. Effects of transfusion on immune function. *Cancer recurrence and infection. Arch Pathol Lab Med.* 1994;118(4):371-379.
46. Waymack JP, Gallon L, Barcelli U, et al. Effect of blood transfusion on immune function. Alterations in macrophage arachidonic acid metabolism. *Arch Surg.* 1987;122(1):56-60.
47. Kirkley SA. Proposed mechanisms of transfusion-induced immunomodulation. *Clin Vaccine Immunol.* 1999;6(5):652-657.
48. Jensen LS, Anderson AJ, Christiansen PM, et al. Postoperative infection and natural killer cell function following blood transfusion in patients undergoing elective colorectal surgery. *Br J Surg.* 1992;79(6):513-516.

