ARAVIND
EYE CARE SYSTEM

Sterilization and Aseptic Technique in Ophthalmic Operating Theatres

Tuesday, January 28th, 2025 edition
DRAFT

Table of Contents

0. Introduction

From April of 2023 to March of 2024, the Aravind Eye Care System (AECS) performed a total of 729,187 surgical procedures across 14 hospitals. This manual describes the techniques that AECS employs to prevent postoperative infections in their operating theatres. Everything from the tools used in surgery to the air allowed into the operating theatre can serve as a means of infection and thus jeopardize the vision of patients. Because the responsibility of upholding strict aseptic technique rests on all members of a hospital's staff, the contents of this manual include duties that will be carried out by personnel in a number of hospital departments.

Although cataract surgery accounts for 90% of the cases of postoperative endophthalmitis (POE), POE poses a significant risk in any ophthalmic surgery. Those who contract this infection often have a grave prognosis on visual outcome, with 20% experiencing severe visual loss. Toxic anterior segment syndrome (TASS) poses a different, but still significant threat to postoperative outcomes. Proper instrument sterilization and the removal of inflammation-causing contaminants in irrigating solutions, medications, and water can reduce the likelihood of this syndrome developing.

Several chapters of this manual describe the reuse of certain surgical devices deemed "single-use" by their manufacturers. This process enables Aravind and other hospital systems in developing nations to keep their procedures low-cost and accessible to their patient populations. "Single-use" devices are not designed or validated for reprocessing (cleaning, disinfection, and sterilization) and run the risk of being degraded over time during this process.

The successes of the aseptic techniques of the Aravind Eye Care System (AECS) have been documented in leading medical journals. A prospective cohort study published in the Journal of Cataract and Refractive Surgery (AG Shulka et al. 2024) found no bacterial or fungal growth in extensive microbiological cultures of 3333 samples collected from instruments destined for reuse at Aravind Eye Hospital, Pondicherry.

AECS does not endorse the reuse of single-use surgical items. The protocols described in this manual are dynamic and likely to evolve as more data is gathered and biomedical advancements are made. It is up to each hospital to evaluate the costs and benefits of these protocols before employing them in practice. The Aravind Eye Care System is not liable for any unintended consequences of the protocols described herein.

1. Aseptic Technique for Hospital Personnel

The skin of the hands has the potential to transfer two unique types of microorganisms: transient and resident microorganisms.

Transient microorganisms: microorganisms that are found on superficial layers of the skin and ARE NOT part of our natural flora. These microorganisms are most easily transferred to others but CAN be easily removed by routine hand hygiene using soap or an alcohol-based sanitizer.

Resident microorganisms: found on and below the superficial layers of the skin (including fingernails, sweat glands, and hair follicles). Although these microorganisms have an important role in the body, they can pose a risk to patients. These microorganisms CANNOT be as easily removed and require an antimicrobial scrub to do so.

Because hands are the most frequent route of transmission of pathogens in hospitals, hand hygiene is critical to both everyday clinical practice (so called **handwashing**) and surgical preparation (so called **surgical scrub**).

1.1 Handwashing and application of hand rub

MATERIALS

- Liquid antiseptic soap OR alcohol-based hand rub▼
- Paper towels

▼ ***NOTE!*** Alcohol-based hand rub is not sufficient on hands that are visibly dirty, contaminated with bodily fluids, or suspected to have been in contact with enteric pathogens (e.g. C. Difficile).

Perform handwashing or apply hand rub when:

- Hands are visibly dirty or contaminated with bodily fluids (*handwash only*)
- After using the toilet or consuming a meal
- Prior to putting on sterile gloves AND after their removal
- After contact with any bodily fluids, mucous membranes, non-intact skin and wound dressings (*handwash only*)
- When entering AND exiting a clinical environment
- When moving from a contaminated body site to a clean body site during patient care
- After handling contaminated laundry, waste, or patient belongings (*handwash only*)
- After cleaning equipment and surroundings

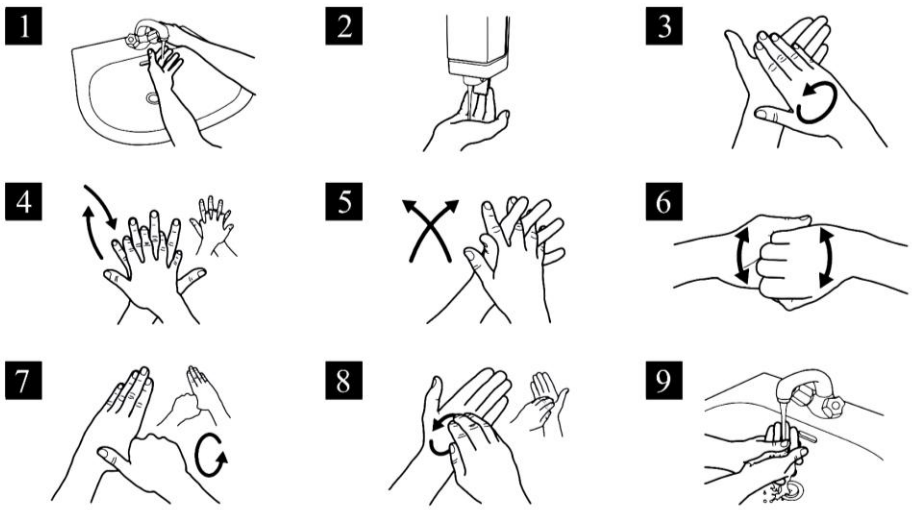
Do not begin hand hygiene unless:

- Cuts and abrasions are covered by a waterproof dressing
- Clothing does not drape below the elbow
- Nails are unpainted and short
- Hand and wrist jewelry has been removed

▼ ***NOTE!*** This procedure should last 40 - 60s. If working with a highly infectious patient, hand hygiene should last 120s prior to contact and 60s following contact.

Handwashing and application of hand rub

1. Wet hands thoroughly in a running tap
    a. If using alcohol-based hand rub, skip this step
2. Apply a palmful of soap (OR alcohol-based hand rub) into the palm
3. Beginning rubbing palm-to-palm to coat all surfaces
4. Keeping both hands with the palm facing down, place the right hand over the left hand and rub with the fingers interlaced
    a. Repeat with the left hand on top
5. Place hands palm to palm and rub with the fingers interlaced
6. Clasp hands and rub the backs of fingers to the opposing palm
7. Clasp the left thumb in the right hand and rub in a circular motion
    a. Repeat with the right thumb
8. Rub the clasped fingers of the right hand in the palm of the left hand in a circular motion
    a. Repeat with the fingers of the left hand
9. Rinse hands with water before drying thoroughly with a paper towel
    a. If using alcohol-based hand rub, skip this step



Images courtesy of the World Health Organization, 2009.

Drying is necessary to prevent microorganism transfer. No communal towels or hot air dryers may be used for this purpose. Paper towels should be of good quality and placed far enough from sinks to avoid contamination by splashes.

1.2 Surgical scrub

MATERIALS
- Liquid chloroxylenol IP 4.8%
- Chlorhexidine gluconate 4% OR povidone-iodine 10%
- Sterile dry towel

If the hands are visibly dirty or known to be contaminated by bodily fluids, **handwashing** using liquid antiseptic soap should be carried out before beginning the surgical scrub.

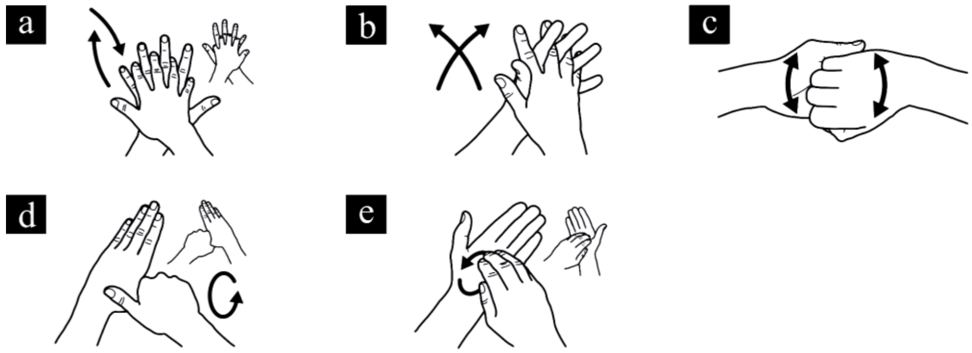
Do not begin hand hygiene unless:
- Cuts and abrasions are covered by a waterproof dressing▼
- Clothing does not drape below the elbow
- Nails are unpainted and short
- Hand and wrist jewelry has been removed
- Dangling accessories which may contaminate the sterile field (e.g. lanyards or stethoscopes) have been removed

▼ ***NOTE!*** Personnel with rashes, open wounds, or bleeding abrasions should scrub or enter the operating theatre.

[SURGICAL SCRUB]

1. Hold the hands higher than the elbows so that water flows downward and drains off the elbows
2. Wet hands thoroughly in a running tap
3. Dispense an appropriate amount of **chloroxylenol IP 4.8%** into the palm and wash from fingertip to two inches above the elbow for approximately 1 minute
    a. Keeping both hands with the palm facing down, place the right hand over the left hand and rub with the fingers interlaced
        i. Repeat with the left hand on top
    b. Place hands palm to palm and rub with the fingers interlaced
    c. Clasp hands and rub the backs of fingers to the opposing palm
    d. Clasp the left thumb in the right hand and rub in a circular motion
        i. Repeat with the right thumb
    e. Rub the clasped fingers of the right hand in the palm of the left hand in a circular motion

i. Repeat with the fingers of the left hand



Images courtesy of the World Health Organization, 2009.

4. Using approximately 5 mL of **chlorhexidine gluconate 4%** OR **povidone-iodine 10%** and sterile water, repeating the previous scrubbing procedure (3) up to two inches above the elbow of each arm
5. Repeat this scrubbing procedure (3) for a third time on but DO NOT advance above the elbow
6. Pass the hands and arms through a stream of warm sterile water in the direction of fingertip to elbow

▼ ***NOTE!*** If sterile water is not available, boiled and cooled water can be used. Boiled and cooled water must be poured by a non-sterile staff member.

7. Keep the hands up above the elbow and away from the body when entering the operating theatre
8. Grasp one end of a sterile dry towel with one hand and dry the opposite arm from the fingers to elbow, only traveling away from the fingers
   a. Repeat for the other arm

The aforementioned scrub procedure should be done before the first case of the day, as well as in between any subsequent cases. At the conclusion of a case, dispose of sterile gloves before re-scrubbing.

1.3 Surgical gloving, gowning, and attire

MATERIALS

- Non-contaminated scrub trousers and shirt
- Disposable or cloth (sterilized) surgical cap
- Disposable or cloth (sterilized) face mask
- Shoe coverings or theatre slippers
- Surgical gown
- Sterile gloves

Following a full surgical scrub, personnel should don a surgical gown and sterile gloves. Personnel must be wearing non-contaminated scrub trousers and shirt, with the shirt tucked into the waistband of the trousers. All personnel should wear a well-fitting surgical cap and shoe coverings or theatre slippers. A face mask should be worn that covers the nose AND mouth.

[GOWNING]

1. Begin by picking up the gown from the table by the exposed inside top layer at the neckline
2. Lift the gown upward and away from the table, allowing for the gown to unfold naturally
3. Slip both hands into the open arm holes while keeping the hands at shoulder level
4. The gown should be adjusted over the shoulder and tied by non-sterile circulating staff
5. Push through the sleeves and allow the hands to extend past the opening
6. Cinch and tuck the cuff strings so that the sleeves fit tightly around the wrist
7. Pass the front tie to the non-sterile circulating staff to be tied
8. The inside or back of the gown should never be touched by the wearer

▼ ***NOTE!*** The inside or back of the gown are unsterile and should never be touched by the hands of the wearer.

[GLOVING]

1. Apply alcohol-based hand rub, following the technique described in section 1.2, *Hand hygiene*
2. Open the glove packet so that the right glove faces the right hand
3. Using the tips of the right fingers, pick up the left glove by its folded inner cuff and lay it over the opposite sleeve with glove fingers facing the elbow and the folded glove cuff resting on the gown cuff
4. Grasp the upper edge of the glove and stretch the cuff of the glove over the hand, flipping the glove to the correct orientation
5. Advance the fingers into the glove
6. Repeat this technique with the opposite hand

▼ ***NOTE!*** Gloved hands should always remain above the navel. Gloves MUST be changed immediately if they touch an object outside of the sterile field. Change gloves after EVERY procedure.

When it is time to dispose of the operating theatre attire, the gown should be removed first before the gloves.

Gown removal

1. Circulating staff should manually untie the knot and pull the gown downward from the shoulder
2. With the outside folded inward, the gown can be collected to be laundered and subsequently autoclaved before reuse

Glove removal

1. Grab the outer surface of one glove cuff and withdraw the hand
2. Using the now exposed hand, grab the inner surface of the other cuff and peel it off the hand, careful to avoid contact with the outer surface of the glove
3. Discard the gloves and any remaining surgical attire

2. Perioperative Patient Asepsis

MATERIALS

- Fresh hospital gown
- Shoe coverings or theatre slippers
- Surgical cap
- Ofloxacin 0.3% eye drops
- Liquid antiseptic soap
- Betadine (povidone-iodine 10%) topical solution
- Povidone-iodine 5% eye drop
- Proparacaine hydrochloride 0.5%
- Moxifloxacin 0.5% intracameral
- Cotton ball and forceps

Careful patient management during the perioperative period is paramount to a successful procedure. Patients should first be appropriately clothed and prepared for the operating theatre. A fresh hospital gown is ideal to minimize contamination of the operating theatre, but a clean outfit will otherwise suffice. Shoe covers (or theatre slippers) and a surgical cap should be provided for the patient. The operative eye should be unambiguously marked on the patient before the surgery begins.

Patients should first be thoroughly evaluated for signs of infection prior to the surgery.

If there is a significant disparity in visual function between the eyes or the patient has had a known recent infection of the eyelid, a conjunctival culture should be performed. If there is any evidence of an ACTIVE infection of the eyelid margin or the conjunctiva (e.g. congestion, hordeolum externum, blepharitis, or discharge), surgery should be postponed.

▼ ***NOTE!*** The following aseptic guidelines are designed specifically for phacoemulsification and manual-small incision cataract surgery. Guidelines for other procedures may vary slightly.

- **On the day prior to surgery,** patients should receive prophylactic **ofloxacin eye drops** administered as directed by the prescribing physician
- **On the day of surgery,** patients should wash their face thoroughly around the eyes and eyelashes using **liquid antiseptic soap**
- **In the preoperative area,** the periorbital region of the operative eye should be painted with betadine (**povidone iodine 10%**) using a cotton ball clamped by forceps
    - Strokes should be made from the medial canthus to the lateral canthus

    - This painting should be repeated once the patient is on the operative table
    - A drop of **proparacaine hydrochloride 0.5%** should be administered into the operative eye
    - A drop of **povidone iodine 5%** should be administered into the operative eye
- **If a block is to be given**, staff should wear a cap, mask, and gloves and use only sterile instruments
- **Intracameral moxifloxacin prophylaxis** should be administered intraoperatively▼
- **At the conclusion of surgery**, one drop of **povidone-iodine 5%** should be installed before the eye is bandaged
- Patients should be provided appropriate antibiotic and steroid drops to be used postoperatively as directed by the prescribing physician

▼ ***NOTE!*** An analysis of 617,453 cataract surgeries performed from January 2014 to May 2016 at the 10 regional Aravind eye hospitals found that routine IC moxifloxacin prophylaxis **reduced the overall endophthalmitis rate 3-fold for manual small-incision cataract surgery and nearly 6-fold for phacoemulsification.**

A Haripriya, DF Chang, RD Ravindran, Endophthalmitis Reduction with Intracameral Moxifloxacin Prophylaxis: Analysis of 600 000 Surgeries, Ophthalmology, Volume 124, Issue 6, 2017, Pages 768-775, ISSN 0161-6420.

3. Sterilization of Surgical Instruments and Devices

It is first useful to define the concept of **sterilization.**

Sterilization is the process of DESTROYING all living microorganisms and their spores. Instruments and tools that are directly used in surgery are required to be sterilized for patient safety. Although this can be achieved by a variety of means, sterilization for medical use is primarily done with steam or chemicals.

3.1 "In-between" instrument sterilization

MATERIALS

- Steel basins
- Soft toothbrush
- 10cc syringe(s)
- 3-line indicators
- Sterilization Chemical Integrator Strip
- Operating theatre attire (including cap, mask, gown, and gloves)
- High-speed ("flash") autoclave

If there are not enough surgical instrument sets to accommodate all scheduled procedures in a day, it will be necessary to perform "in-between" sterilization using a high-speed autoclave so that sets can be reused within the same day. "In-between" sterilization will begin with manual cleaning followed by a cycle in the high-speed autoclave. Some specialty instruments may have specific "in-between" sterilization measures, as described in Chapter 4, *Specialty Instrument Cleaning Protocols*.

Undergo basic hand hygiene and dress in **operating theatre attire** (including cap, mask, gown and gloves) before instrument cleaning of any kind.

"In-Between" Instrument Cleaning

1. Dispose of the visco cannula, capsulotomy needle, simcoe syringe, and disposable knives
2. Fill two basins with sterile water
3. Dump the contents of a surgical tray into basin #1 and wash thoroughly with a **soft toothbrush** in order to remove all debris, tissue, and bloodstain
4. Rinse the trays in basin #2
5. Using a **10cc syringe**, flush each of the cannulated items into basin #3
6. Place each cleaned instrument and item back into the cleaned tray

Flash autoclave preparation

1. Place a **3-line indicator** sticker into each tray
2. Pack the washed instruments/trays perpendicularly into a clean bin
3. Place a **sterilization chemical integrator strip** near the center of the bin

3.2 "End of day" instrument cleaning

MATERIALS

- Steel basins
- Soft toothbrush
- 20cc syringe(s)
- 5cc syringe(s)
- Sterile cloth and drape
- Clean hand cloth
- Operating theatre attire (including cap, mask, and gloves)
- Steam autoclave
- Ultrasonic cleaner
- Hair dryer
- Tip covers

Begin by preparing a work area. Clean the surface of the work area with sterile water and cover with an appropriately sized sterile cloth and drape. Undergo basic hand hygiene and wear operating theatre attire (including cap, mask, and gloves) during instrument cleaning of any kind. (Fig 1)

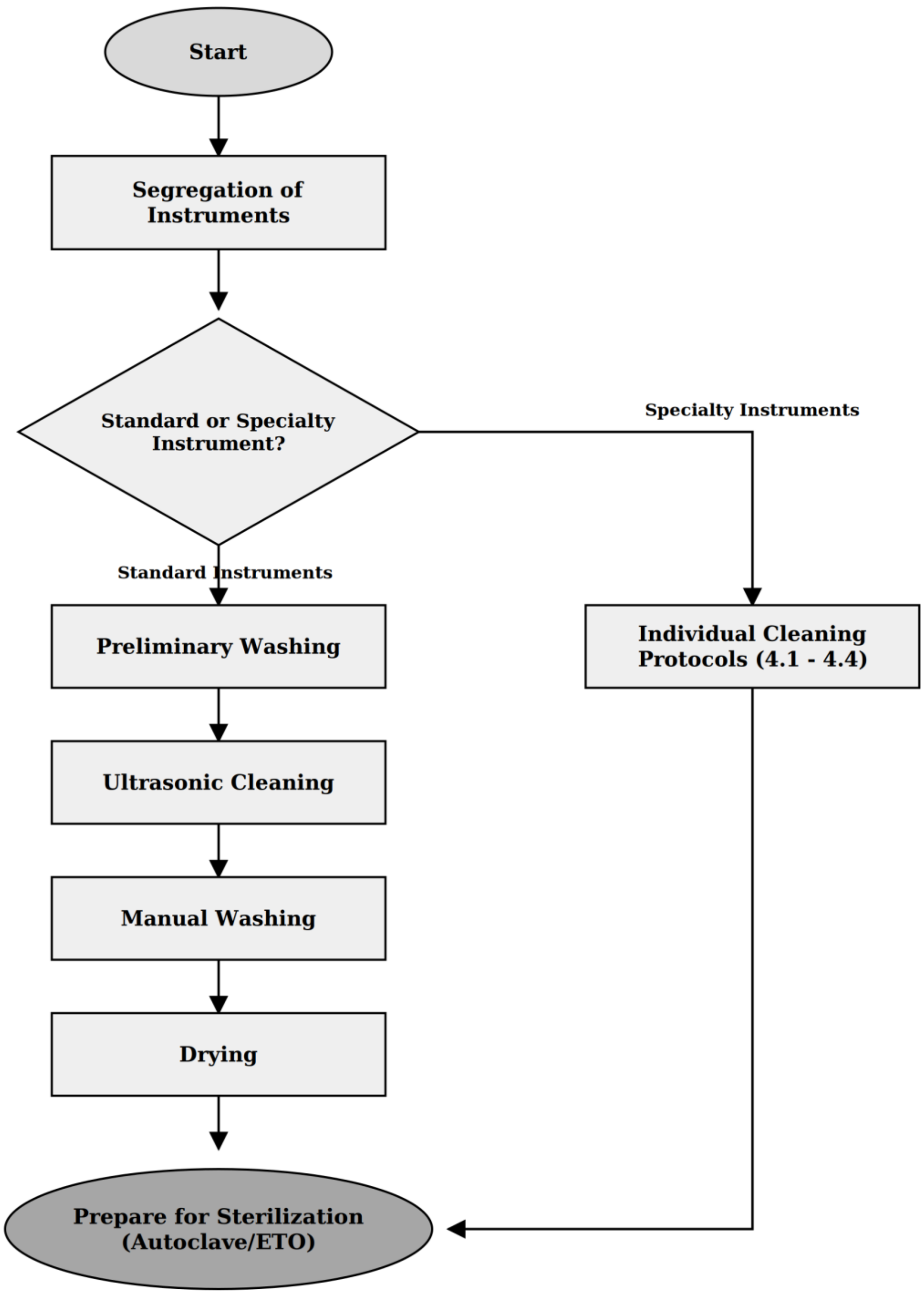
Certain specialty instruments will have individual cleaning protocols prior to sterilization which are described in Chapter 4, *Specialty Instrument Cleaning Protocols*. These instruments are as follows:

Cataract	Phaco handpiece, I/A handpiece, hydro cannula, vitrectomy probe, 4-0 silk thread, superior rectus needle, knife (crescent, keratome, side port), rhexis needle, Simcoe cannula/tube, iris hook, phaco tip/sleeves/chamber, I/A tip
Retina	Vitrectomy cutter, endo-illumator/endo-laser probe, diathermy wire/tip, maxgrip forceps/scissors/endo-scissors, oil tube/needle, gas bottle, cryoprobe
Cornea	Corneal trephine/punch, 3mm DMEK punch, Lieberman teflon block, DSAEK punch, Moria 350 head, Geuder glass cannula, Barron artificial anterior chamber, vitrectomy probe, cryoprobe
Glaucoma	Diode probe, capsule hook

Segregate the standard instruments into separate set trays containing each of the following:

- Blunt instruments
- Sharp instruments
- Cannulated items

Fig 1: Preparation for sterilisation



Preliminary Washing

1. Immerse all instruments of a given set into a basin and wash thoroughly with a soft toothbrush in order to remove all debris, tissue, and blood stain
   a. Clean inside the jaws of all hinged instruments
   b. Clean complex instruments with many surfaces (e.g. speculums, needle holders) multiple times
   c. Flush all cannula thoroughly
2. Return the instruments to the set tray
3. Repeat with a fresh basin for the remainder of the set trays

Ultrasonic cleaning

1. Submerge each set tray in the tank of a cleaned ultrasonic cleaner filled with enough sterile water so that the water level is one inch above the top of the tray(s)
2. Switch on the machine for a three minute cycle
3. Repeat this cycle once for cannulated instruments
4. For each cycle, drain and refill the ultrasonic cleaner with fresh sterile water

▼ ***NOTE!*** If ultrasonically cleaning multiple set trays in the same cycle, ensure that the trays do not overlap.

Manual cleaning

1. Immerse all instruments in basin #1 and wash thoroughly with a soft toothbrush in order to remove all debris, tissue, and bloodstain
   a. Clean inside the jaws of all hinged instruments
   b. Clean complex instruments with many surfaces (e.g. speculums, needle holders) multiple times
2. Rinse all instruments in basin #2
3. Rinse all instruments in basin #3
4. Wipe instruments with a clean hand towel and carefully inspect for damage or leftover buildup
5. Begin to place items onto the drape, creating separate stacks for each instrument
6. Allow all instruments approximately 5 minutes to air dry

▼ ***NOTE!*** Hinged instruments should dry in the “open” position.

7. Evaporate residual water with a hair dryer
8. Place fresh (unused) tip covers on sharp instruments

Specialty instrument cleaning

Certain items will require specific cleaning measures prior to sterilization. Refer to chapter 5, *Specialty Instrument Cleaning Protocols*.

3.3 Sterilization using steam

MATERIALS

- Clean cloth
- Clean hand towels
- 3-line label indicators
- Aluminum cap opener
- Black/white light chamber
- Autoclave tape
- Sodium hypochlorite
- Cloth bundling material
- Autoclave bins

Preparing an instrument set tray

1. Lay out cleaned and dried instrument set trays on a clean cloth
2. Place a clean hand towel into each instrument set tray
3. Begin placing items into the tray, including all instruments necessary for a particular surgery in a given tray
   a. If applicable, place small specialty items (i.e. simcoe/hydro/visco cannula) inside a small cup within each tray
4. Place a **3-line label indicator** inside the tray to ensure adequate steam penetration
5. Fold the ends of the towels toward the middle of each tray, covering the instruments

Preparing used and open bottles

1. Open the aluminum cap of the used bottles using an opener
2. Clean BOTH bottle and corks in tap water
3. Place the bottles in a light chamber with black and white backgrounds
4. Shake the bottle under the light to detect any dust or particles
   a. If the bottle is completely empty, fill with mineral water and close with corks before placing into bin as described
5. Holding the bottle in the black portion, check for white particles or cracks
6. Holding the bottle in the white portion, check for dark particles
7. Clean the bottle thoroughly and dry with a clean cloth
8. Place groups of 25 bottles in a bin with a sterile towel at the bottom
9. Place autoclave tape in a folded sterile towel and place it at the top of the bin

Preparing other items for the autoclave

- **Linens:** all linens must first be soaked in **sodium hypochlorite** solution and laundered before being bundled in cloth with like items and equipped with indicator stickers at the bottom, middle, and top of the bundle
    - **Eye towels** should be folded, keeping the stitched eye hole visible and bundled in groups of 15
    - **Table towels** should be folded as small as possible and bundled in groups of 15
    - **Gowns** should be knotted and folded in a way in which the inside portion will face the surgeon when opened and bundled in groups of 6
    - **Small towels** should be bundled in groups of 20
- **Eye shields:** clean and dry using sterile water and a fresh cloth, place into a prepared autoclave bin with appropriate indicator tape

Preparing autoclave bins

1. Cover the inner surface of the cleaned bin with a clean dry towel
2. Place a 3-line label indicator at the bottom of the bin
3. Begin placing trays perpendicular to each other to enable optimal steam penetration
4. Place one 3-line label indicator and one cloth-wrapped Sterilization Chemical Integrator Strip at the center of the packed bin
5. Place one final 3-line label indicator at the top of the bin
6. Place two more 3-line label indicators at the middle and top of the packed bin
7. Place one sterilization chemical integrator strip in the center

Note that **instruments** and **liquids** should never be placed in the same autoclave load.

The exact procedure of running the autoclave will depend on the exact model used. The following conditions should be met to ensure sufficient steam penetration:

Item	Pressure	Temperature	Time
Instruments and linen	20 lbs	121°C	30 minutes
Bottles	20 lbs	121°C	20 minutes

Some general precautions are applicable no matter what autoclave model is used:

- Change the sterile water in the machine daily
- Close the bin holes immediately after the cycle ends
- Ensure that all indicators confirm sterilization

Following each autoclave cycle, retain the following items in their own designated register or file:

- Autoclave process printout (including date, program run, temperature, pressure, and total sterilization time
- All 3-line label indicators used in the cycle
- The sterilization chemical integrator strip used in the cycle

Proper storage of sterilized items is crucial to maintaining sterility. Items should remain in a clean and dry environment and should only be handled when necessary, by masked personnel.

3.4 Sterilization using ethylene oxide

MATERIALS

- Cotton pads
- ETO sterilization pouches
- Sealing machine and medi peel paper
- Cutting machine or scissors
- 3-line label indicators
- Sterilization chemical integrator strip
- ETO machine and trolley
- Ethylene oxide gas cartridge

Ethylene oxide (ETO) can be used to sterilize items that are heat or moisture-sensitive, and thus cannot go through traditional steam sterilization. ETO kills all known viruses, bacteria, fungi, and spores by interfering with the normal metabolism of proteins. Due to its possible hazards, ETO should only be used to sterilize items that CANNOT undergo steam sterilization.

Packaging for ETO sterilization

- Cleanse the instruments with a sterile water dampened cotton pad and remove any detachable parts
- Allow all instruments to dry COMPLETELY and place into ETO sterilization pouches pouches
- Flatten the ETO sterilization pouches pouch to remove all air and seal using the sealing machine
- Paste a 3-line label indicator▼ on the outside of the pouch
- Place the ETO sterilization pouches pouch into the cleaned ETO sterilizer, ensuring that items do not touch the walls or ceiling of the chamber
- Double pack a Sterilization Chemical Integrator Strip and place it into the center of the ETO sterilizer before running the cycle

▼ ***NOTE!*** Sterilized products should be appropriately labeled with the operator’s name, sterilization date, and expiry date (1 month for single pack, 6 months for double packs). Reusable items should include a log of the number and dates of reuse, ensuring that reuse is never in excess of what is recommended.

The exact procedure of running the ETO sterilizer will depend on the exact model used. The average sterilizing cycle will take **approximately 7.5 hrs.** Ensure all indicators within a cycle confirm sterilization before any of the items are used.

Following each autoclave cycle, retain the following items in their own designated register or file:

- ETO sterilizer process printout (including date, program run, temperature, pressure, and total sterilization time
- All 3-line label indicators used in the cycle
- The sterilization chemical integrator strip used in the cycle

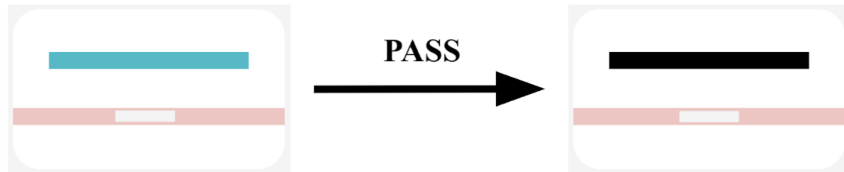
Store sterilized items in the sterile storage room and away from direct sunlight. This room should be kept at 40-60% relative humidity and 22°C to 26°C.

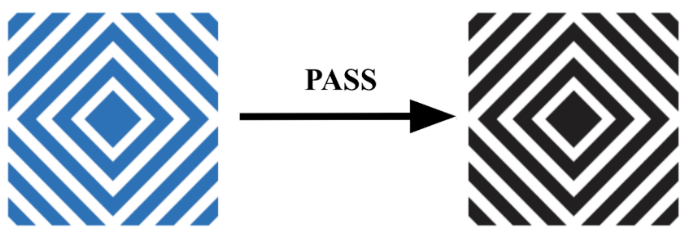
3.5 Sterilization indicators

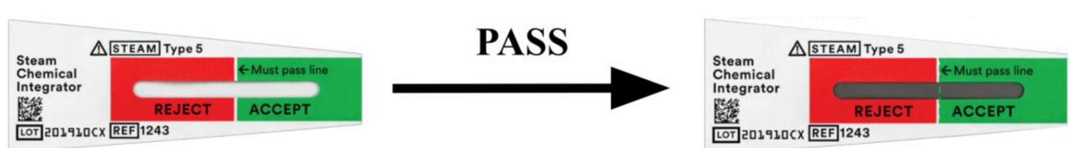
Indicators are useful tools to ensure that sterilization has been achieved properly.

Classes of indicators

- Class (**I**): “Process indicators” distinguish between items that have been sterilized and those which have not
- Class (**II**): “Test indicators” confirm proper functioning of the autoclave
- Class (**IV**): “Multi-parameter indicators” indicate exposure to a sterilization cycle at certain values of two or more chosen parameters (e.g. temperature and steam time)
- Biological indicators (**BI**): indicate whether necessary conditions are sufficient enough to kill spores through the use of highly resistant microbes

<table>
<tr><th>Indicator</th><th>Frequency of usage</th><th>Color change</th><th>Indicator Type</th></tr>
<tr><td>3-Line Indicator</td><td>Every bin/load/set</td><td>Varies</td><td>Class I</td></tr>
<tr><td colspan="4">


BLUE BLACK
Use instructions
Paste indicator label on ALL items that are to be sterilized. For bundles, stick three labels throughout the bundle (top, middle, bottom). For instruments, stick a label in each instrument tray. Label with the following information: sterilizing personnel, autoclave load #, bin and bundle # (line 1); date of sterilization (line 2); expiration date (line 3). Maintain a register containing the outcome of each 3-line indicator used. The indicator ink is specific to either autoclave or ETO sterilization. Verify the correct indicator to use before starting a cycle.
Look for: uniform color change
</td></tr>
<tr><td>Bowie-Dick</td><td>Weekly</td><td>Varies</td><td>Class II</td></tr>
<tr><td colspan="4">


BLUE BLACK
Use instructions
Before the first load of the week, place the Bowie-Dick-type test sheet in the center of the test pack. Place horizontally on the floor of the sterilizer rack, near the door and over the drain in an otherwise empty chamber. Initiate the test with three pre-vacuum pulses before reaching the 132.2°C set point. Maintain a register containing the results of each Bowie-Dick test run.
Look for: uniform color change
</td></tr>
<tr><td>Sterilization Chemical Integrator Strip</td><td>Daily (Every bin/load)</td><td>White → Black</td><td>Class IV</td></tr>
<tr><td colspan="4">


</td></tr>
</table>

<table>
<tr><td colspan="4">Use instructions
Place indicator labels on ALL items that are to be sterilized. Do so in the location of the package deemed to be LEAST accessible to sterilization gas or steam. Maintain a register containing the outcome of each sterilization chemical indicator strip used. Ensure that steam integrator strips are placed with items which will be autoclaved and ETO integrator strips are placed with items that will be sterilized by ETO gas.
Look for: intrusion of black pigment into the “ACCEPT” window</td></tr>
<tr><td>Steam Indicator</td><td>Weekly</td><td>N/A</td><td>BI</td></tr>
<tr><td colspan="4">

Use instructions
Seal one BI indicator into a ETO sterilization pouches pack weekly and place into the autoclave along with other items. Run the sterilization cycle as normal. Send the indicator-pack to the microbiology department. Maintain a register containing the outcome of each Steam BI.
Look for: acceptable organism growth according to indicator manufacturer instructions</td></tr>
<tr><td>ETO Indicator</td><td>Weekly</td><td>N/A</td><td>BI</td></tr>
<tr><td colspan="4">

Use instructions
Seal one BI indicator into a ETO sterilization pouches pack weekly and place into the ETO sterilizer along with other pouches. Run the sterilization cycle as normal. Send the indicator-pack to the microbiology department and verify acceptable organism growth. Maintain a register containing the outcome of each ETO BI.
Look for: acceptable organism growth according to indicator manufacturer instructions</td></tr>
</table>

▼ ***NOTE!*** Indicator failures should be appropriately documented, reported, and investigated. Items cannot be used until sufficient evidence of sterility.

4. Specialty Instrument Cleaning Protocols

The reuse of certain surgical devices deemed "single-use" by their manufacturers enables Aravind and other hospital systems in developing nations to keep their procedures low-cost and accessible to their patient populations. "Single-use" devices are not designed or validated for reprocessing (cleaning, disinfection, and sterilization) and run the risk of being degraded over time during this process.

The successes of the aseptic techniques of the Aravind Eye Care System (AECS) have been documented in leading medical journals. A prospective cohort study published in the Journal of Cataract and Refractive Surgery (AG Shulka et al. 2024) found no bacterial or fungal growth in extensive microbiological cultures of 3333 samples collected from instruments destined for reuse at Aravind Eye Hospital, Pondicherry.

The instruments and devices described below are reused in some capacity for a variety of the subspecialty services offered at AECS. Items should always be discarded if they show any signs of damage or malfunction.

AECS does not endorse the reuse of single-use surgical devices. It is up to each hospital to evaluate the costs and benefits of these protocols before employing them.

4.1 Instruments used in cataract surgery

▼ ***NOTE!*** Be careful to keep track of the number of times a given instrument is reused, such as by writing the date of sterilization and the number of times it has been used on the outside of its ETO pouch.

Item	# of Reuses	Method	Requirements
Phaco handpiece	Until damaged	Autoclave	20cc syringe

Reprocessing instructions

Immediately following each surgical procedure, flush irrigation and aspiration ports 10x with a 20cc syringe filled with warm sterile water. Dry by flushing both ports with an empty 20cc syringe. Pack into an instrument tray and autoclave with the appropriate indicators.

▼ ***NOTE!*** Not all phaco handpiece models should be washed by aspiration. Refer to the manual provided by the manufacturer for detailed cleaning instructions.

<table>
<tr><td>I/A handpiece</td><td>Until damaged</td><td>Autoclave</td><td>20cc syringe</td></tr>
<tr><td colspan="4">Reprocessing instructions
Immediately following each surgical procedure, flush irrigation and aspiration ports 10x with a 20cc syringe filled with warm sterile water. Dry by flushing both ports with an empty 20cc syringe. Pack into an instrument tray and autoclave with the appropriate indicators.
▼ NOTE! Not all I/A handpiece models should be washed by aspiration. Refer to the manual provided by the manufacturer for detailed cleaning instructions.</td></tr>
<tr><td>Hydro cannula</td><td>Until end of day</td><td>Autoclave</td><td>Basins (3), 5cc syringes</td></tr>
<tr><td colspan="4">Reprocessing instructions
Fill two basins with sterile water and keep one basin empty. Wash the instrument by hand in basin #1. Using water from basin #2, flush the cannula into basin #3 with a 5cc syringe. Flush the cannula once with air using an empty 5cc syringe. Run through a high-speed autoclave cycle once completely dry.</td></tr>
<tr><td>Vitrectomy probe</td><td>10</td><td>ETO</td><td>Saline, basins, ETO pouch</td></tr>
<tr><td colspan="4">Reprocessing instructions
Turn on the Visco mode on the phaco machine. Put the probe into a basin filled with saline. Pressing the foot pedal, wash the probe until the bowl is empty. Dry the probe with a sterile wiper. Close the vitrectomy tip with the provided cap and seal the probe into an ETO pouch. Run through an ETO sterilization cycle.</td></tr>
<tr><td>4-0 silk thread</td><td>Until end of day</td><td>Autoclave</td><td>Basin</td></tr>
<tr><td colspan="4">Reprocessing instructions
Place the thread into its own instrument tray. Take 1 basin filled with sterile water and clean any blood residue by hand. Run through a high-speed autoclave cycle once completely dry.</td></tr>
<tr><td>Superior rectus needle</td><td>Thread: end of day
Needle: end of week</td><td>Autoclave</td><td>Basin</td></tr>
<tr><td colspan="4">Reprocessing instructions
Seperate the needle from the thread. Take 1 basin filled with sterile water and clean any blood residue from the needle. Lightly dry with a hand towel. Run through a high-speed autoclave cycle once completely dry. At the end of the day, clean the needle using a 3 minute ultrasonic cleaner cycle before cleaning with a basin of sterile water and autoclaving.</td></tr>
<tr><td>Knife (Crescent, keratome, side port blade)</td><td>5</td><td>Autoclave</td><td>Basin, soft toothbrush</td></tr>
</table>

<table>
<tr><td colspan="4">Reprocessing instructions
Take 1 basin filled with sterile. Remove the cap on the knives and wash both sides gently. Run through a high-speed autoclave once dry.</td></tr>
<tr><td>Rhexis needle</td><td>Until end of day</td><td>Autoclave</td><td>Basins, 5cc syringes</td></tr>
<tr><td colspan="4">Reprocessing instructions
Fill two basins with sterile water and keep one basin empty. Wash the instrument by hand in basin #1. Using water from basin #2, flush the cannula into basin #3 with a 5cc syringe. Flush the cannula once with air using an empty 5cc syringe. Run through a high-speed autoclave cycle once completely dry.</td></tr>
<tr><td>Simcoe cannula and tube</td><td>Cannula - until blockage
Tube - end of week</td><td>Autoclave</td><td>Basins, 5cc syringe</td></tr>
<tr><td colspan="4">Reprocessing instructions
Between patients, fill two basins with sterile water and keep one basin empty. Wash the instrument by hand in basin #1. Using water from basin #2, flush the cannula into basin #3 with a 5cc syringe TWICE. Flush the cannula once with air using an empty 5cc syringe. Run through a high-speed autoclave cycle once completely dry.

At the end of day, fill two basins with sterile water and keep one basin empty. Wash the instrument by hand in basin #1. Using water from basin #2, flush the cannula into basin #3 with a 5cc syringe FIVE TIMES. Clean the entire apparatus with compressed air. Run through a high-speed autoclave cycle once completely dry.</td></tr>
<tr><td>Iris hook</td><td>10</td><td>ETO</td><td>Sterile tray, 5cc syringe, 27g cannula, banyan cloth, ETO pouch</td></tr>
<tr><td colspan="4">Reprocessing instructions
Fill a sterile tray with sterile water. Fill a 5cc syringe with sterile water and attach the 27g cannula. Wash the hook with the force of the water using the syringe and cannula FIVE TIMES. Wrap into banyan cloth and allow to dry. Place in the designated iris hooks box. Place cleaned and dried hooks into individual ETO sterilization pouches. Run through an ETO sterilization cycle.</td></tr>
<tr><td>Phaco tip, sleeves, chamber, I/A tip</td><td>Until damaged</td><td>Autoclave</td><td>Basins, 20cc syringes</td></tr>
<tr><td colspan="4">Reprocessing instructions
Following ultrasonic cleaning, fill one basin with sterile water. Flush the cannulated items FIVE TIMES using a sterile-water-filled 20cc syringe. Dry the lumen by flushing FIVE TIMES with an empty 20cc syringe and allow the exterior to air dry. Pack into an instrument tray and autoclave with the appropriate indicators.</td></tr>
</table>

4.2 Instruments used in retina surgery

<table>
<tr><th>Item</th><th># of Reuses</th><th>Method</th><th>Requirements</th></tr>
<tr><td>Vitrectomy cutter</td><td>25g - 3
23g - 6</td><td>ETO</td><td>Irrigating solution (Glutasol @ 650), sterile wet wipes, compressed air, ETO pouch</td></tr>
<tr><td colspan="4">Reprocessing instructions
Immediately following use, rinse the probe using the irrigating solution via aspiration. Aspirate air to dry the lumen of the probe. Open the adaptor and aspirate irrigating solution followed by air for 1 minute.

At the end of day, clean the cutter surface using wet wipes and dry using compressed air. Pack and sterilize via ETO.</td></tr>
<tr><td>Endo-illuminator/Endo-laser probe</td><td>6</td><td>ETO</td><td>Sterile wet wipes, compressed air, ETO pouch</td></tr>
<tr><td colspan="4">Reprocessing instructions
Clean the surface of the illumination probe with sterile wet wipes and dry using compressed air. Pack and sterilize via ETO.</td></tr>
<tr><td>Diathermy wire and tip</td><td>10</td><td>ETO</td><td>Fresh diathermy tip, sterile wet wipes, ETO pouch</td></tr>
<tr><td colspan="4">Reprocessing instructions
Change the diathermy tip following each case. Clean with a sterile wet wipe, dry, and pack for sterilization via ETO.</td></tr>
<tr><td>Maxgrip forceps/scissors/endo-scissors</td><td>6</td><td>ETO</td><td>Sterile wet wipes, compressed air, fresh “reusable” tip, ETO pouch</td></tr>
<tr><td colspan="4">Reprocessing instructions
Immediately following a procedure, put the forceps in a closed tray containing sterile water.

At the end of day, clean with a sterile wet wipe and dry using compressed air. Change the “reusable” tip after each case and pack for sterilization via ETO.</td></tr>
<tr><td>Oil tube and needle</td><td>Tube - 12
Needle - 6</td><td>ETO</td><td>Sterile wet wipes, compressed air, ETO pouch, fresh needle</td></tr>
<tr><td colspan="4">Reprocessing instructions
During surgery, clean the needle by flushing with sterile water.

Between patients, change the needle.</td></tr>
</table>

At the end of day, flush the needle with sterile water before ultrasonically cleaning for 6 minutes. Flush with sterile water, dry, and pack for ETO or steam sterilization. **At the end of day,** extract and inject sterile water with the oil tube 10 times. Ultrasonically clean for 6 minutes. Wipe with wet wipes and dry using compressed air.			
Gas bottle	Until empty or expires	ETO	Sterile wet wipes, ETO pouches
Reprocessing instructions After using the gas quantity required, clean the exterior of the container with sterile wet wipes and dry using compressed air. Pack for ETO sterilization.			
Cryoprobe	Until deformation	ETO	Sterile wet wipes, compressed air, ETO pouches
Reprocessing instructions **In between surgeries,** clean the exterior with sterile wet wipes. **At the end of day,** clean the exterior with sterile wet wipes and dry using compressed air. Pack for ETO sterilization.			

4.3 Instruments used in cornea surgery

Item	# of Reuses	Sterilization	Requirements
Corneal trephine & punch	Until rust or deformation	ETO	Sterile tray, cotton wiper, ETO pouches
Reprocessing instructions Wash the trephine and punch in a sterile tray filled with sterile water. Clean the inner and outer surfaces using a wet cotton wiper. Allow both items to air dry and examine for rust or damage. Pack separately for ETO sterilization once fully dry.			
3mm DMEK punch	4 (after initial use)	ETO	Sterile tray, 20cc syringes, cotton pad, ETO pouch
Reprocessing instructions Wash the punch in a sterile tray filled with sterile water. Flush the punch once using sterile water in a 20cc syringe through the rear end of the punch. Clean the outer surface using a wet cotton pad. Flush 5 times with an empty 20cc syringe. Pack for ETO sterilization once fully air dried.			
Leiberman teflon block	Until deep grooves form	ETO	Sterile tray, cotton wiper, ETO pouch

Reprocessing instructions Wash in a sterile tray filled with sterile water. Clean the surfaces of the block using a wet cotton wiper. Pack for ETO sterilization once fully air dried.			
DSAEK punch	3 (after initial use)	ETO	Sterile tray, cotton wiper, cotton pad, ETO pouch
Reprocessing instructions Wash the punch in a sterile tray filled with sterile water. Clean the inner and outer surface of the metallic portion of the punch using a wet cotton wiper. Clean the surface of the plastic portion of the punch using a clean wet cotton pad. Pack for ETO sterilization once fully air dried.			
Moria 350 head	4 (after initial use)	ETO	Sterile tray, 20cc syringe, hydro cannula, banian cloth, ETO pouch
Reprocessing instructions Wash the head in a sterile tray filled with sterile water. Flush the cutting portion of the head once with a sterile water-filled 20cc syringe with hydro cannula attached. Flush the head once more with a sterile water-filled 20cc syringe (without the cannula). Dry with a clean banian cloth and pack for ETO sterilization once fully dried.			
Geuder glass cannula	Until damaged or cracked	ETO	Sterile tray, 5cc syringes, banian cloth, ETO pouch
Reprocessing instructions Wash the cannula in a sterile tray filled with sterile water. Flush the cannula FIVE times using a 5cc syringe and sterile water. Dry the inner lumen by flushing TEN times using an empty 5cc syringe. Dry the outer surface with a clean banian cloth and pack for ETO sterilization once fully dried.			
Barron artificial anterior chamber	5 (after initial use)	ETO	Basin 5cc syringe, cotton pad, ETO pouch
Reprocessing instructions Flush each cannula FIVE times with sterile water into an empty basin. Dry the inner lumen of each cannula by flushing it TEN times using an empty 5cc syringe. Dry the outer surface with a clean cotton pad and pack for ETO sterilization.			
Vitrectomy probe	10	ETO	Saline, basins, ETO pouch
Reprocessing instructions Turn on the Visco mode on the phaco machine. Put the probe into a basin filled with saline. Pressing the foot pedal, wash the probe until the bowl is empty. Dry the probe with a sterile wiper. Close the vitrectomy tip with the provided cap and seal the probe into an ETO pouch. Run through an ETO sterilization cycle.			
Cryoprobe	Until damaged	ETO	Sterile wet wipes, compressed air, ETO pouches

Reprocessing instructions
In between surgeries, clean the exterior with sterile wet wipes. **At the end of day,** clean the exterior with sterile wet wipes and dry using compressed air. Pack for ETO sterilization.

4.4 Instruments used in glaucoma surgery

Item	# of Reuses	Method	Requirements
Diode probe	5	ETO	Sterile tray, cotton pad, ETO pouch
Reprocessing instructions Wash the probe in a sterile tray filled with sterile water. Clean the tubing and tip of the probe with a wet cotton pad, always moving from the distal to the proximal end. Allow to air dry and pack for ETO sterilization once completely dry.			
Capsule hook	10	ETO	Banyan cloth, iris hooks box, forceps
Reprocessing instructions Wash the probe in a sterile tray filled with sterile water. In case of visible blood stains, hold the hook with forceps and wash the hoop with a forceful spray of sterile water using a syringe and cannula. Wrap in banyan cloth and allow to dry fully. Place dry hooks into the iris hooks box using forceps. Pack the hooks box for ETO sterilization.			

5. Aseptic Technique in the Operating Theatre

The operating theatre environment should be managed to maximize sterility and always kept free of clutter. If the operating theatre is used for general (non-ophthalmic procedures), the ophthalmic procedures for the day should be scheduled first. In the event that the ophthalmic surgery is being conducted on an infected person, the operating room may not be used again until it is thoroughly cleaned and fumigated.

5.1 Cleaning of the operating theatre

Cleaning should be performed to varying degrees on a DAILY, WEEKLY, and TRIMONTHLY basis. This cleaning should always begin in the operating theatre itself, before progressing toward the scrub area, preoperative and postoperative areas, and the sterilizing room in that order. The date and extent of each cleaning should be documented in an OT cleaning register.

Daily cleaning

Object(s) to be cleaned	Cleaning product & method	Cleaning instructions
Operating tables, trolley, surgeon’s chair, foot stool, walls, and floor	**1% Microl Solution** (50mL Microl - F and 5L sterile water)	Clean first using sterile water followed by 1% Microl Solution application. Sweep the floors with a soft broom before cleaning. The Microl Solution application should be done twice for the walls and the floor.
All other operating theatre furniture, phacoemulsification machine	Sterile water	Wipe using a clean cloth wetted in sterile water
Microscope	**Isopropyl alcohol** or **alcohol-based handrub solution**	Wipe using a clean cloth, avoiding **ALL** optic components
Scrub sink and transportation carts	**1% Microl Solution** (50mL Microl - F and 5L sterile water)	Clean first using sterile water followed by 1% Microl Solution application.
Autoclaves and ETO sterilizers	Varies	Clean according to manufacturer-provided instructions

Ultrasonic cleaner	Antiseptic liquid concentrate (15% Cetrimide and 3% Chlorhexidine gluconate)	Disinfect first with antiseptic liquid concentrate before rinsing with sterile water and drying suing a clean cloth

Weekly cleaning▼

▼ ***NOTE!*** ALL mobile objects should be removed from the operating room prior to the weekly cleaning procedure

Object(s) to be cleaned	Cleaning product and method	Cleaning instructions
Air-conditioning unit	Sterile water and **1% Microl Solution** (50mL Microl - F and 5L sterile water)	Remove the filter and scrub with a toothbrush and sterile water. Dip the filter into the 1% Microl Solution and wipe the inner spaces of the unit.
Operating theatre table, suction holders, sitting stools, footstools, stands, IV poles, basin stands, X-ray view boxes, metal trolleys, oxygen tank hoses, kick buckets, and wall cupboards	**Surf powder solution,** clean tap water, and **instruclean.**	Thoroughly scrub with surf powder solution, wash with clean tap water, and wipe with instruclean and a fresh cloth **after** drying
Floor and walls	**Surf powder solution** and clean tap water	Scrub with surf powder solution and allow to sit for five minutes before washing with clean tap water
Ceiling	**Microl - F**	Remove any ceiling cobwebs using a soft brush, thoroughly wipe with a Microl - F soaked cloth

Doors, knobs, hinges, handles, and glass inserts	**Bacillocid 0.5%**	Wipe with a bacillocid moistened cloth.
Stains (if present)	**Scrubbing machine**	Follow manufacturer-provided instructions
Waste buckets	**Benzalkonium chloride (BAC) 5%**	Sterilize with BAC and rinse with water

Follow weekly cleaning with fumigation of the operating theatre. Use a clean fumigator machine and equal parts **Formalin 40%** and sterile water. Follow manufacturer-provided instructions and wear proper PERSONAL PROTECTIVE EQUIPMENT including a respirator when handling this carcinogenic solution. Ensure that the room is sealed, the air-conditioning unit is off, and no personnel enters the operating theatre until at least twelve hours have passed. Document this fumigation in an OT fumigation register.

Trimonthly cleaning

- The operating theatre should be thoroughly water-washed once every three months
- Fumigation and a bacterial culture (*see section 6.1*) should be performed after this cleaning.

5.2 Water and air in the operating theatre

Air in the operating theatre can act as a medium for infection if it is not managed carefully. The air conditioning system should be designed to facilitate several principles:

- Airflow should go from the area directly surrounding the operating table, to the outer operating theatre and adjacent rooms, to the unsterile staff areas
- Outside air should not be allowed to directly enter the operating theatre
- Window & split air conditioning should be avoided
- Positive pressure inside of the operating theatre should be maintained at all times
- Maintenance should be carried out on a regular basis (often every 6 months) to maintain proper system status

To prevent microbial growth, the operating theatre should be maintained at a temperature of 22° - 26°C and a humidity of 45% - 60%. Both metrics should be checked each morning and recorded in a temperature and humidity register. See *section 6.1* for air culturing details.

Air Metrics to Be Achieved

Air Changes Per Hour	Minimum of 20, with a fresh air change a minimum of 4 out of a total 20 changes
Air Velocity	25-35 ft/min from a non-aspirating unidirectional laminar flow array
Positive Pressure	Minimum of 2.5 Pascal
Air Filtration	Two sets of filters of efficiency 90% (down to) 10 microns and 99% (down to) 5 microns

An adequate supply of reverse-osmosis filtered water must be kept at all times. The water level, cleanliness, and chemical dosing record should be checked each morning. See *section 6.1* for water culturing details.

6. Surveillance and Documentation

A crucial step in preventing postoperative endophthalmitis and toxic anterior segment syndrome is regular evaluation of the aseptic techniques and sterilization cycles being carried out. Microbial surveillance analyzes the microbial flora in the operating theatre environment, instruments, and consumables to inform as to whether steps personnel ensure that requisite sterilization procedures are adequate. Documentation and record keeping creates a log which allows personnel to ensure that requisite aseptic protocols and sterilization procedures are being performed when required.

6.1 Microbial Surveillance

Ringer lactate surveillance

1. TWO DAYS BEFORE using a new batch of ringer lactate bottles, send one bottle for pH testing and culturing
2. The pH can be adjusted if it is not ideal (6.5)
    a. If < 6.5 and > 6.0: add NaOH (sodium hydroxide) to basify
    b. If > 6.5 and < 7.0: add HCl (hydrochloric acid) to acidify
3. Discard any batch where pH values are considered abnormal or culture reports are not satisfactory
4. Maintain a file containing all ringer lactate culture reports

Vicomet surveillance

1. TWO DAYS BEFORE using a new case of viscomet, send one bottle for pH testing and culturing
2. Discard any batch where pH values are considered abnormal or culture reports are not satisfactory

Water surveillance

1. EVERY 15 DAYS fill three 30mL bottles with approximately 22.5mL of the following water sources:
    a. Raw water
    b. Reverse-osmosis (RO) water
    c. Reverse-osmosis (RO) water with capsule filter
2. Perform cultures and check for the following conditions:

Raw water	Less than 500 microbial colonies
RO water	Less than 100 microbial colonies
RO water with capsule filter	No microbial colonies

3. Fungus, *Pseudomonas* bacteria, or *E. coli* bacteria are **unacceptable** in any water source
4. Maintain a file containing all water culture reports

> ▼ ***NOTE!*** If the culture report is not satisfactory, ensure that proper action is taken the cultures are repeated and confirmed acceptable before the water is used in any clinical or surgical setting.

Air surveillance

1. EVERY MONTH obtain the following air samples inside of the OT
    a. One culture plate near the door of the operating theatre
    b. One culture plate on either side of the operating theatre
    c. One culture plate held beneath the AC unit for 20 minutes
    d. One culture plate prepared from EACH the microscope handle, instrument trolley, and operation table
2. Perform cultures and check for the following conditions for each culture plate

Number of bacterial colonies	Less than 20
Number of fungal colonies	None

3. Maintain a file containing all air culture reports

> ▼ ***NOTE!*** If the culture report is not satisfactory, ensure that proper action is taken and the cultures are repeated and confirmed acceptable before operating theatre is used.

When new saline batches are received, the bottles should be examined for high integrity and sampled for a microbiology culture before being transferred to the operating theatre. **In the case of Hepatitis-B or HIV positive patients**, cases should be performed at the end of the day. Specialized personal protective equipment should be worn and burned following surgery.

6.2 Documentation and record keeping

Registers are exceptionally useful in maintaining accurate records of the sterilization process and aseptic procedures. The number of registers and the form they take are likely to vary between hospitals but certain topics are universally applicable.

Recommended registers

Register subject	Content
Indicators	● Test result (pass/fail) ● Date of test and performing personnel ● Next due date (if on recurring basis) ● Identifying information about the machine, cycle, and parameters (if relevant)
Culture reports	● The raw microbial values obtained ● The date and location of culturing ● Test result (pass/fail) ● Steps being taken to rectify (if failed)
Sterilization printing files	ETO: ● Exposure and aeration time of each cycle ● Record of successful completion Steam: ● Program run ● Temperature and pressure reached ● Total sterilization time per cycle
Recalls	● The item that was recalled ● Identifying information about the sterilization cycle, operator, date, and parameters ● Details of the repeating sterilization cycle
Staff meetings	● Time and date of meeting (should happen weekly) ● Issues discussed ● Preventative measures agreed upon

7. Training and Continuing Education

Successful implementation of aseptic technique and sterilization protocols requires thorough training of all staff involved. A comprehensive training program should be devised by senior hospital faculty in order to teach staff how to execute their required duties properly.

The following sterilization-related processes should be observed by all staff during training:

- Cleaning of instruments for “in-between” sterilization
- Cleaning of instruments for “end-of-day” sterilization
- Cleaning of linens, eye shields, and bottles
- Bin packing and unpacking
- Documentation in the registers
- Indicator usage and microbial culturing
- Recall procedure
- Operating theatre cleaning (end of day and weekly)

The following operating theatre processes should be observed by all staff during training:

- Set-up of the OT trolley
- Surgical scrub procedure
- Gowning and gloving
- Preoperative patient preparation
- Patient preparation by the assistant (including draping)
- Postoperative patient preparation

It is the responsibility of the senior hospital faculty to evaluate the readiness of all staff to participate in sterilization protocols and aseptic technique. Staff should be given regular opportunities to participate in continuing education in order to refresh their knowledge and adapt to new policies or protocols.

i. Materials Master List

1. Aseptic Practices for Hospital Personnel	● Liquid antiseptic soap ● Alcohol-based hand rub ● Paper towels ● Chloroxylenol IP 4.8% ● Chlorhexidine gluconate 4% OR povidone-iodine 10% ● Reverse osmosis purified warm sterile water ● Sterile dry towel ● Non-contaminated scrub trousers and top Disposable or cloth (sterilized) surgical cap ● Disposable or cloth (sterilized) face mask ● Shoe coverings or theatre slippers ● Surgical gown ● Sterile gloves
2. Perioperative Patient Asepsis	● Fresh hospital gown ● Shoe covers ● Disposable or cloth (sterilized) surgical cap ● Disposable or cloth (sterilized) face mask ● Ofloxacin 0.3% eye drops ● Liquid antiseptic soap ● Betadine (povidone-iodine 10%) topical solution ● Proparacaine hydrochloride 0.5% ● Povidone-iodine 5% eye drop ● Intracameral moxifloxacin 0.5% ● Cotton ball and forceps
3. Surgical Product Asepsis	● Steel basins ● Soft toothbrushes ● 10cc syringe(s) ● 3-line indicators ● Sterilization Chemical Integrator Strip ● Operating theatre attire (including cap, mask, gown, and gloves) ● High-speed (“flash”) autoclave ● 20cc syringe(s) ● 5cc syringe(s) ● Sterile cloth and drape ● Clean hand cloth ● Steam autoclave **(such as Nat Steel 24 SR/DD Autoclave)**

	● Ultrasonic cleaner ● Hair dryer ● Sharp instrument tip covers ● Aluminum cap opener ● Black/white light chamber ● Autoclave tape ● Sodium hypochlorite ● Cloth bundling material ● Autoclave bins ● Cotton pads ● ETO sterilization pouches ● Sealing machine and medi peel paper ● Cutting machine or scissors ● ETO machine and trolley ● Ethylene oxide gas cartridge ● Bowie-Dick-type test sheets ● Steam biological indicators ● ETO biological indicators
4. Specialty Instrument Cleaning Protocols	● 20cc syringe(s) ● 5cc syringe(s) ● Basins ● Saline ● ETO sterilization pouches ● Soft toothbrushes ● Sterile tray(s) ● 27g cannula ● Banyan cloth Irrigating solution (Glutasol @ 650) ● Sterile wet wipes ● Compressed air ● Cotton wipers ● Cotton pads ● Hydro cannula ● Iris hooks box ● Forceps
5. Aseptic Practices for the Operating Theatre	● Microl - F ● Soft broom ● Clean cloth ● Isopropyl alcohol OR alcohol-based handrub solution

	• Antiseptic liquid concentrate (Cetrimide 15% and Chlorhexidine gluconate 3%) • Surf powder solution • Bacillocid 0.5% • Scrubbing machine (such as **Roots scrubbing machine**) • Benzalkonium chloride (BAC) 5% • Fumigator machine • Formalin 40% • Respiratory PPE
6. Microbial Surveillance of the Operating Theatre and Surgical Products	• Sodium hydroxide • Hydrochloric acid • 30mL bottle(s) • Culture plate(s) (soybean casein digest agar)