Arizona State University
Institutional Animal Care and Use Committee
STANDARD INSTITUTIONAL GUIDELINE

RODENT SURGERY

It is the policy of the IACUC that all survival surgeries involving rodents be conducted using appropriate aseptic techniques as described in this guideline.

A. Aseptic Technique

1. General Information
   a. Although mice and rats have been touted as being resistant to post-surgical infections, existing literature documents how certain subclinical infections can influence study results or become clinical diseases following stress or immune suppression (Cooper et al, 2000).
   b. In view of this information, rodent survival surgery must be performed using aseptic technique, the primary objective of which is to reduce microbial contamination of the incision and exposed tissues to the lowest possible practical level.
   c. Aseptic technique must be used in the preparation of the surgical surface, the animal, the surgeon, and the surgical instruments.

2. Preparation of the Surgical Surface
   a. Before beginning rodent surgery, the laboratory bench or table where the surgery will be performed should be cleaned and disinfected. Quaternary ammonium compounds (e.g., Virex) or Accel are good choices for such disinfection.
   b. Laboratory benches in front of open windows, next to doors, or similar locations where air currents and dust are difficult to control should be avoided as rodent surgery tables. Likewise, rodent surgery should not be performed in or in front of an exhaust hood unless it is required for biosafety.

3. Preparation of the Animal
   a. Enough fur should be shaved from the incision site to prevent wound contamination, but, because of the small size of rodents, care should be taken not to remove excessive amounts of fur so as to interfere with the animal's thermoregulation.
   b. The incision site should be disinfected first with either a dilute iodine solution (e.g., Betadine) or dilute chlorhexidine (e.g., Nolvasan), followed by 70% alcohol. This process should be repeated two additional times. The disinfectant should not be washed or rinsed from the surgical site.
   c. Each step in the disinfectant-alcohol process should be applied in a concentric fashion beginning at the incision site and moving outward, away from the incision site to the periphery of the surgical field.
4. Preparation of the Surgeon

a. All individuals participating in surgery should be wearing a face mask and hair cap.

b. It is recommended that all individuals performing surgery wash their hands before donning gloves. Recommended agents include an iodine scrub (such as Betadine scrub) or a chlorhexidine scrub (such as Nolvasan scrub).

c. All individuals performing rodent surgery should wear sterile surgical gloves. Alternately, exam gloves wiped with hydrogen peroxide and peracetic acid or with 70% isopropyl alcohol have been shown to be nearly as effective (Keen et al., 2010; LeMoine et al., 2015). Disinfecting gloves can be particularly suitable if the surgeon’s hands never touch sterile materials or internal body tissues. Exam gloves can also be autoclaved (LeMoine et al., 2015).

5. Preparation and Use of Surgical Instruments

a. Surgical instruments for rodent surgery should be sterilized. Steam or gas sterilization is optimal, but hot bead sterilization is more practical and equally effective when only instrument tips will contact the animal. While inferior because it is not a sterilant, 70% isopropyl alcohol can be effective at preventing bacterial contamination (Huerkamp, 2002; Keen et al., 2010), and may, with IACUC approval, be approved for some situations (e.g., field surgery on multiple animals without a source of electricity).

b. Rodent surgeries are oftentimes done several in succession. In such cases, it is imperative that instruments are cleaned and sterilized between surgeries. Between animals, instrument sterilization is best achieved using a hot bead sterilizer, but instruments should be cooled to room temperature prior to contacting the animal. Cooling can be expedited by soaking the sterilized instruments in sterile water, alcohol, or equivalent solution. Approval for using a chemical disinfectant (e.g., quaternary ammonium compounds, iodine solutions, alcohol, and phenol compounds) instead of a hot bead sterilizer must be pre-approved by the IACUC based on rationale as to why a hot bead sterilizer cannot be used.

c. Once sterilized, parts of the instruments that will touch internal tissue must only contact sterile surfaces. During surgery, but when not in use, instrument tips should be kept on a sterile surface or suspended so that they do not touch any surface. Realize that, when using bead sterilization, the handles of the instruments are not sterile, so instrument tips should not come in contact with the handle of another instrument or touch a place where an instrument handle has previously been. If the tip of an instrument touches a non-sterile surface, it should be re-sterilized.

d. It is recommended to either use one pair of scissors and forceps for skin incisions and a second pair of scissors and forceps for abdominal wall incisions and manipulation of viscera or to disinfect instruments in a hot bead sterilizer after opening the body wall.

e. To avoid contamination of sterile items, it is recommended that the animal’s body, other than the site of the incision, be covered with a sterile drape or sterile gauze. Clear plastic wrap (e.g., Saran Wrap) can be used to cover the body to avoid gross contamination of the surgical site with hair or dander, but, as the wrap is not sterile, contact of it with sterile instruments must be avoided.
f. If internal tissue needs to be exteriorized through the incision and will re-enter the body cavity, it must be placed on a moist sterile surface (e.g., a piece of sterile gauze dampened with sterile saline).

B. Facility

According to the Guide for the Care and Use of Laboratory Animals, a separate facility specifically designated for rodent surgery is not required. The Guide states that, “for most rodent surgery, a facility may be small and simple, such as a dedicated space in a laboratory appropriately managed to minimize contamination from other activities in the room during surgery.”

C. Choice of Surgical Materials

1. Expired drugs cannot be used for any surgery and expired supplies cannot be used on animals that will recover from surgery.

2. Surgical Instruments

The size of the rodent must be considered when planning a surgical procedure. The surgical instruments (scalpel blade, scissors, forceps, etc.) need to be matched to the size of the rodent and the procedure in order to minimize trauma and maximize success. The use of proper instruments and surgical technique are likely as important as the use of aseptic technique in preventing post-surgery infections (Cooper et al., 2000).

3. Surgical Needles

a. Cutting needles should be used when it is necessary to suture dense, difficult to penetrate tissue such as skin.

b. Non-cutting needles (such as taper point or round needles) are used primarily for suturing easily penetrated tissues such as peritoneum or intestine. Use of cutting needles on fine tissue will readily tear the tissue during the placement or the suture or, worse, upon recovery.

4. Suture Materials, Surgical Staples, and Suture Patterns

a. Absorbable Sutures

(1) Absorbable suture materials should be used on internal tissue when the animal is expected to survive long-term. It can also be used for skin closure if there is limited tension on the incision. Absorbable suture is preferred for field studies to avoid the need to recapture the animal or permanently leave sutures in.

(2) Examples of absorbable suture material appropriate for rodent surgery include Vicryl, Monocryl, and PDS.

b. Non-absorbable Sutures

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(1) Non-absorbable suture materials should be used whenever tension exists on an incision and the potential for dehiscence exists. Non-absorbable suture materials are especially appropriate for skin closure provided the suture can be removed in 7-14 days.

(2) Monofilament non-absorbable suture material is recommended for skin closure to avoid possible wicking of contaminants into the wound. However, monofilaments can be more difficult to work with and have greater memory than braided suture.

(3) Examples of non-absorbable suture material appropriate for rodent surgery include nylon and Prolene.

c. Surgical Staples

(1) Surgical staples are an effective and time-efficient alternative to suture material for skin closure.

(2) Surgical staples should be removed 10-14 days post-surgery, as their extended presence can cause irritation. To avoid skin damage, staples are best removed using a purposefully designed staple remover.

d. Suture Patterns

(1) To avoid detrimental outcomes (e.g., internal hemorrhage, skin dehiscence), suturing skill must be well established prior to working on a live animal that is expected to recover from the procedure.

(2) Improperly tied knots can unravel, so care must be taken to use proper suture technique to assure knot security.

(3) The abdominal wall of an animal, and other high-tension critical tissues should be closed using a simple interrupted pattern rather than a single continuous running suture. This reduces the probability that a large defect will occur should a knot or tissue placement fail. The same is true for skin closure, but to a lesser extent since the skin has less tension, is less vulnerable to tearing, and can be visually inspected to detect problems early.

(4) Rodents can chew sutures out, so it is advisable to use a buried suture pattern (e.g., a running subcuticular suture pattern) or staples when closing the skin.

D. Anesthesia

Consult the Anesthesia SIG for acceptable choices of rodent anesthetic agents.

E. Post Surgical Care

Consult the Post-operative Surgical Care SIG for appropriate post-operative monitoring following rodent surgery.
F. Analgesia

Consult the Analgesia SIG for acceptable choices of rodent analgesic agents.

G. References:


