Background:

The Federal Animal Welfare Act Regulations and the *Guide for the Care and Use of Laboratory Animals* 8th Edition (*Guide*) both set standards that are obligatory for biomedical research involving live, vertebrate animals. These documents have been adopted by the Association for the Assessment and Accreditation of Laboratory Animal Care, International (AAALAC) and virtually all public and private funding sources, and are therefore pertinent to all research projects, regardless of funding source. The following guidelines augment statements contained in the *Guide* and further define rodent surgery guidelines pertaining to UCSB.

I. Surgery Facilities

- The *Guide* states: “For most survival surgery performed on rodents and other small species such as aquatics and birds, an animal procedure laboratory, dedicated to surgery and related activities when used for this purpose and managed to minimize contamination from other activities conducted within the room at other times, is recommended.”¹ In the ideal world, there should not be any other activities occurring in the research laboratory when rodent surgeries are being performed, but this is not practical, and may not be necessary. The UCSB IACUC agrees with AAALAC’s assessment: “AALAC International acknowledges that limiting nonsurgical activities in the laboratory may help to minimize contamination of the surgical area. However, AAALAC recognizes that minimizing contamination during surgery may be achieved by considering several factors. The specific location of the surgical area within the laboratory should promote the proper conduct of sterile technique, and to the extent possible, it should be isolated from other activities in the laboratory. The surgical area should be dedicated for that purpose while surgery is performed. Other factors that may impact the risk of contamination include the invasiveness and complexity of the surgical procedure, duration of surgery, and the nature of other non-surgical activities conducted in the laboratory (i.e. their likelihood of increasing the risks of surgical contamination). For complex or long procedures, or if the layout of the laboratory does not permit a suitable dedicated surgical space, it may be advisable to temporarily stop other laboratory activities, thereby dedicating the laboratory to surgery in order to maximize the potential
for a good surgical outcome. For minor surgeries of short duration, conducted in a suitable area within the laboratory, it may be acceptable to allow other laboratory activities to continue if they do not jeopardize sterile technique. The investigator, IACUC (or comparable oversight body) and veterinarian should evaluate surgical areas to ensure they are appropriate.

- The area where surgeries are performed should be uncluttered to minimize the potential for contamination and facilitate ease of cleaning. The surfaces (e.g. tables) in the surgical area should be impervious to moisture (e.g. stainless steel). The surgical area should be routinely cleaned and disinfected.

II. Surgeon Training and Qualifications

- Personnel conducting surgical procedures on the species being maintained or studied will be appropriately qualified and trained in those procedures.\(^1\),\(^2\) This is required to ensure that good surgical technique is practiced including asepsis, gentle tissue handling, minimal dissection of tissue, appropriate use of instruments, effective homeostasis, and correct use of suture materials and pattern.\(^1\)

- All surgeons must be specifically identified on the animal protocol, and they must be qualified and trained to perform surgical procedures as defined by the IACUC guideline, *Training Requirements for Protocol Personnel*. Qualifications and training shall be listed on the IACUC Protocol Training Roster.

- The Campus Veterinarian should be consulted for questions regarding animal health, surgical wound care, animal anesthesia techniques, surgical procedures, and provision of post-operative care. Additionally, surgical outcomes should be continually and thoroughly assessed to ensure that appropriate procedures are followed and timely corrective changes are instituted.\(^1\) The Campus Veterinarian should be consulted when there is an increased incidence of anesthetic or peri-operative deaths, or increased incidence of post-surgical complications.

- The following training video in the Journal of Visualized Experiments is recommended: *Principles of Rodent Surgery for the New Surgeon*

III. Preparation of Surgical Instruments

- Surgical instruments should be sterilized for use in survival and non-survival rodent surgery. Several techniques (steam, dry heat, ethylene oxide, or chemical agents) can be used to sterilize instruments and other materials that will come in contact with the animal's tissues. The sterilization method selected will depend on the physical characteristics of the materials to be sterilized. For example, autoclaving is the preferred methods to sterilize common surgical instruments. By contrast, delicate instruments (micropipettes or cannulas) may be damaged by steam or dry heat sterilization
procedures, and therefore should be sterilized by immersion in liquid chemical sterilants or high-level disinfectants. If chemical sterilants are used, then manufacturer’s recommendations on contact times should be followed, and the material must be rinsed in sterile water before use. Alcohol (i.e. ethyl alcohol or isopropyl alcohol) is neither a sterilant nor a high-level disinfectant, and therefore should not be used to sterilize surgical instruments. However, alcohol at concentrations between 60-90% is rapidly bactericidal against vegetative forms of bacteria, and a potent viracidal and fungicidal agent. Its use should be restricted to applications were the presence of bacterial spores is not anticipated, for example, decontamination of autoclaved surgical instruments between serial rodent surgeries.

- Following sterilization, surgical instruments and medical or surgical devises (e.g. catheters, cannulas, or osmotic mini-pumps) must be handled using aseptic technique to prevent contamination. Further, these sterile instruments and devices must be properly stored. The shelf life of these instruments will depend on how they were prepared and wrapped, how they are stored, and event-related factors contributing to their contamination. Storage of surgical instruments in plastic self-seal sterilization pouches is recommended. Storage in closed or covered cabinets is preferred, but open shelving may be used for storage. Any sterile instrument, device, or sterile package containing these items that has fallen or been dropped to the floor, is exposed to water, or experiences any other event that could contribute to the contamination of the sterile material should be cleaned and re-sterilized.

- When performing surgeries on multiple animals (mice or rats), at least 2 sets of instruments should be sterilized by an appropriate method. While the first set of instruments is being cleaned to remove all organic material (i.e. blood) and re-sterilized, the second set is used on a second animal. If only the tips of instruments have contacted the sterile surgical field or the animal’s organs/tissues, then the instrument tips may be cleaned to remove organic material (i.e. blood), and placed in a glass bead sterilizer for a few seconds (check instrument manufacturer for recommended contact times). As an alternative to using a glass bead sterilizer in tips-only surgical procedures, surgical instruments may be immersed in a sterile bowl containing 70% isopropyl alcohol for at least 2 minutes. The instruments should be maintained in a sterile holder/container after decontamination (glass bead or alcohol), and until used for the next surgery. Decontamination of surgical instruments with alcohol during serial tips-only surgeries must be limited in use, and after the 5th animal, fresh sterile instruments must be used.

IV. Preparation of the Animals

- Prior to taking the animal to the surgery area, remove all hair for at least a centimeter on either side of the incision site. Hair removal should be performed in a location away from the surgery area to minimize the potential for contaminating the surgery area. Hair can be removed by clipping with a #40 clipper blade, shaving with a razor, plucking (in anesthetized mice or similar-sized rodents), or by using a depilatory cream. Vacuum or otherwise remove loose hair.
• Clean and aseptically prepare the surgical site. Alternate between a topical antiseptic solution (e.g. Betadine) and 70% alcohol to carefully clean and disinfect the skin surface. Use a surgical sponge, cotton swab, or cotton-tipped applicator to apply the topical antiseptic solution and alcohol. Clean in a gradually enlarging circular pattern from the center of the proposed incision to the periphery. For surgeries in mice, the surgical site is often very small making the above cleaning technique impractical. In these situations, focus on cleaning the surgical area and avoid bringing the sponge or swab back from the contaminated periphery to the clean surgical area. Repeat the skin preparation cycle for a total of three times, each time beginning at the center and proceeding to the periphery. End the preparation of the surgical site with an application of antiseptic solution (i.e. Betadine).

• To prevent hypothermia, try not to wet the animal any more than necessary.

• Care should be taken to prevent contamination of the sterile surgical field during subsequent handling and positioning of the animal. Glad’s Press’n Seal freezer wrap provides an uncontaminated (i.e. free of microorganisms and organic material), inexpensive and effective method to cover the surgical field.

• Place lubricating ophthalmic ointment (such as Lacrilube or Paralube) on the anesthetized animal's eyes to prevent drying of the cornea.

• Place the animal on a clean absorbent surface (e.g. disposable underpads or blue chuxs) and maintain its body temperature using a water circulating heating pad, or equivalent external heat source, taking care not to overheat the animal.

V. Preparation of the Surgeon

• Surgeons should wash their hands before surgery.

• Surgeons should wear a mask, sterile gloves, and clean scrub shirt or lab coat to prevent contamination of the sterile surgical field. A new pair of sterile surgical gloves should be used for each animal. Alternatively, if only the tips of the surgical instruments will come into contact with the surgical field or animal's organs/tissues, then the same pair of surgical gloves may be decontaminated between animals and used on up to 5 animals. To decontaminate the gloves, either spray them with 70% isopropyl alcohol and rub the wet gloved hands together for 30s, or dip the fingertips of the gloves in a sterile bowl containing 70% isopropyl alcohol for 30s.

VI. During Surgery

• The surgical field must be kept sterile throughout the procedure. Sterile instruments must be prevented from contacting non-sterile surfaces. Catheters, probes, or cannulas must
not contact non-sterile surfaces, for example, fur or paws. **In most cases, sterile drapes are required for maintenance of the sterile field.**

- Monitor and control the animal's body temperature.
  - Use a rectal temperature probe or thermometer, of appropriate size for mice or rats, to monitor the animal's body temperature.
  - Use a water circulating heating pad, or a heating plate/pad, to maintain the animal's normal body temperature: mice (36.5 - 38°C) and rats (35.9 - 37.5°C).

- Monitor and control the level of anesthesia.
  - The exact monitoring procedure and frequency should be based on the invasiveness and complexity of the surgical procedure, and the duration of the surgery. In general, more intensive intraoperative monitoring, both in terms of methodology and frequency, is required for longer duration, technically complex, and invasive surgeries. Similarly, the surgical records should reflect these differences, and will be evaluated by the IACUC to assess the adequacy of the intraoperative monitoring procedures.
  - However, and at a minimum, the surgeon or the anesthetist should pay continuous and close attention to the animal's anesthetic depth by:
    - Checking the animal’s response to painful stimuli (incision, toe/tail pinch) - there should be no response or movement.
    - Checking the animal's respiratory rate (i.e. breaths per minute) and character - it should not be too slow or too shallow
    - Checking the animal's cardiac output and tissue perfusion - the color of mucous membranes should be pink and moist; the color of the eyes (albino rodents) should be red; and/or the color of the skin (ears) should not be pale, or blue.

- Control any intraoperative bleeding by placing a sterile gauze (e.g. 2 x 2) or sterile cotton-tipped applicator over the bleeding area and applying gentle pressure. Blot, do not rub or wipe the area to remove or soak up the blood. Alternatively, sterile absorbable gelatin sponges (e.g. Gelfoam) may be used to control bleeding.

- Use the appropriate material and method to close the surgical incision.
  - Stainless steel **wound clips** are recommended for closing skin incisions in rodents.
  - Tissue adhesive (glue; Vetbond™ or Dermabond™) should only be used for a topical application and only to close easily approximated skin edges (i.e. no tension) created by a sterile surgical incision. Tissue adhesive must be used sparingly (only one or two drops), and applied only on the cut edges of the skin, and not on the deeper tissues. The applicator tip should not contact the tissue, and if it does it should be discarded and replaced. Click on the hyperlink for additional **application tips**.

- Provide analgesic treatment(s) to alleviate the pain caused by the surgical procedure. The evolving standard of veterinary care for rodent surgical procedures is to provide
multimodal analgesic treatments combining a local anesthetic (e.g. lidocaine or bupivacaine) with opioid (e.g. buprenorphine) and/or non-steroidal anti-inflammatory analgesics (e.g. flunixin meglumine or meloxicam) immediately prior to or during surgery, and then to administer additional doses of the analgesic treatments, at prescribed dosing intervals, during the postoperative period. Consult with the Campus Veterinarian regarding analgesic treatment and doses for your specific surgical procedure.

- If an emergency evacuation occurs during surgery, and the animal being operated on has an exposed body cavity (e.g. craniotomy), open incision(s), or exposed cannulas or catheters, then that animal should be euthanized by administering an overdose of anesthesia prior to evacuating the area, if time and circumstances permit. If the animal is anesthetized but no or only minor surgical manipulations have been performed, then leave the anesthetized animal to recover in a confined space (e.g. a cage in the surgery room) and evacuate the area. Employees should never put themselves at risk during an emergency evacuation to care for animals.

VII. Postoperative Care

- The animal should be continuously monitored until it is conscious, or can be aroused when handled, and has stable vital signs (e.g. regular breathing pattern and rate).

- The animal should be recovered from surgery by itself (i.e. not housed in a cage with other animals) on a clean and dry surface (no bedding), and it should be provided with supplemental heat during the recovery period. For example, the animal may be placed on absorbent pad in a clean and empty rodent cage that is resting on a heating pad. Be cautious with supplemental heat sources; hyperthermia can be as detrimental as hypothermia.

- Provide fluid replacement therapy, as needed. Fluid replacement therapy is not usually required for most rodent surgeries, because they do not involve prolonged operative times and cause only minimal bleeding. However, if the animal (rat or mouse) is slow to recover from anesthesia, then provide warmed fluids (sterile saline, or Lactated Ringer’s Solution) at a volume of 10 ml/kg for rats (e.g. 1 ml per 100 gm) or 25 ml/kg (i.e. 0.25 ml per 10 gm). These fluids should be administered subcutaneously.

- Animals should not be returned to the animal room until they can stand and move about their cage. To prevent cannibalism or suffocation, continue to house rodents individually until they can stand and move around. Additionally, continue to house rodents individually even after they are completely recovered from anesthesia, if they have exposed (i.e. not protected by a metal cover) external catheters or ports.

- Make sure that the animal has access to food and water when it's returned to the animal room. If necessary, provide food/water on the bottom of the cage in the form of moistened pellets or Diet Gel.
A member of the research team or other individual to whom postoperative care has been delegated must monitor each animal at least once a day and document all observations and treatments in the postoperative recovery log.

Each animal must be carefully observed for signs of pain, and surgical complications, including infection or dehiscence of the surgical site(s). Pain assessment guidelines are included below.

Surgical wounds (e.g. skin incisions, vascular access ports, and cephalic implants) should receive appropriate wound care (e.g. cleaning skin wounds and cephalic implant skin margins with antiseptic wipes). External wound clips or sutures should be removed 7-14 days after the surgery.

The Campus Veterinarian should be contacted if there are any unexpected complications.

VIII. Pain Management and Assessment

- The fourth principle of the U.S. Government Principles for the Utilization and Care of Vertebrate Animals Used in Testing, Research, and Training states: “Proper use of animals, including the avoidance or minimization of discomfort, distress, and pain when consistent with sound scientific practices, is imperative. Unless the contrary is established, investigators should consider that procedures that cause pain or distress in human beings may cause pain or distress in other animals.”

- Technical proficiency in aseptic surgical technique, especially gentle tissue handling, is very important in reducing pain from surgery, because: "As a rule, pain is likely to occur in proportional terms as a result of tissue injury—more extensive tissue damage results in greater pain and thus a need for a stronger analgesic regimen."

- Please refer to this link for a review of the effects and doses of anesthetic and analgesic drugs tailored to our animal care and use program. In all cases, the anesthetic and analgesic plans for the rodent surgical procedures practiced in our animal care and use program were developed in consultation with the Campus Veterinarian, and were based on the predicted pain intensity of the surgical procedure and the expectation that the surgical procedure will be performed by a member of the research team that is technically proficient in the surgical technique. The anesthetic and analgesic treatment plan is described in the IACUC protocol. It is therefore expected that members of the research team will be technically proficient in the surgical technique and will administer analgesic treatments as described in the approved IACUC protocol.

- Members of the research team are expected to monitor, at appropriate intervals, the effectiveness of analgesic treatments, and notify the Campus Veterinarian if the analgesic treatments are not effectively managing the animals' pain.

- The response of animals to pain, and hence any clinical or behavioral signs of pain, will vary depending on the animal species and the surgical procedure. There is no simple
cage-side evaluation of pain in rodents. A nuanced interpretation of animal behavior is required to make a reasonable assessment of animal pain and distress. The best way to identify signs of pain is to closely observe the appearance and behavior of the animal for at least two days prior to surgery, and note any changes after surgery. However, the behavioral assessment only works well if the animals have a varied repertoire of normal behaviors, which only exists when animals are housed in an enriched environment.

- The following signs are associated with persistent pain in rodents.⁶, ⁷
  - Reduced level of spontaneous activity
  - Reduced grooming
  - Reduced nest building (in mice)
  - Partially closed or squinted eyes
  - Piloerection (hair standing up)
  - Increased aggressiveness or vocalization when handled (in rats)
  - Abnormal posture (hunched posture in mice; arched backed and/or tucked-in abdomen in rats)
  - Reduced food and water intake resulting in rapid weight loss

IX. Records², ⁸

- Records are required for all surgical procedures (survival or non-survival) in order to document the appropriate performance of anesthesia and surgery, and to demonstrate compliance with the surgical, anesthetic/analgesic, and post-operative plan in the approved protocol. The surgery, anesthesia, and postoperative care record should be kept in the room where the animals are housed. Having the record in the room accomplishes several functions. 1) It explains the condition of the animals to animal care staff (a sedated animal may otherwise be thought to be ill). 2) It assures animal care staff and USDA Animal Welfare inspectors that the animal is being cared for. 3) It informs animal care staff how recently the investigator has seen the animal; this knowledge helps them decide whether or not there is a need to contact the investigator to inform him or her of the present condition of the animal.

- Although individual records are desirable, a composite record may be used for a group of rodents. The record (individual or composite) should include the surgery date, name of the surgeon(s), a brief description of the surgical procedure, any drugs or treatments that were administered, and a note of any complications or previous surgical procedures that may have been performed on the animal. The latter is required so that the IACUC or other regulatory agency (e.g. USDA, AAALAC) can assess whether or not an animal has undergone more than one survival surgical procedure.

- The record should document that the animal was appropriately anesthetized prior to making the first surgical incision, and it should identify the intraoperative assessments of the animal's physiological status that were performed at regular intervals (e.g., every 15 minutes). Additionally, it should include notations of any variations in vital signs and anesthetic depth that were observed and actions that were taken to correct such variations.
The record should include a notation for each time the animal was examined postoperatively. After all wounds have healed and all sutures/wound clips have been removed, the postoperative record requires no further entries, but should continue to be kept in the area where the animals are housed.

Clicking on either of the following links will reveal a template for a rodent anesthesia and surgery record, and a postoperative record recommended by the Campus Veterinarian for most rodent surgical procedures in our animal care and use program. Alternatively, the PI may use a different template for their lab, but it should capture and document important information with comparable detail. Please consult with the Campus Veterinarian to ensure that the level of surgical and postoperative documentation in your template is appropriate for your surgical procedure and species.

Reference:

2. American Association for the Assessment and Accreditation of Laboratory Animal Care, International. Frequently Asked Questions
3. Public Health Service Policy on the Humane Care and Use of Laboratory Animals