**Survival Surgery in Rodents and Birds**

**I. Background**

Recommendations for the performance of survival surgery on mice, rats, and birds are based on the 2011 edition of the NIH Guide for the Care and Use of Laboratory Animals (the Guide, pp. 115-119, 144-145) and the USDA Animal Welfare Act Regulations (AWARs) §2.31(d)(1)(ix) and §2.33(a)(5).

Guide: p. 115 (8th ed.): “Successful surgical outcomes require appropriate attention to presurgical planning, personnel training, anesthesia, aseptic and surgical technique, assessment of animal well-being, appropriate use of analgesics, and animal physiologic status during all phases of a protocol involving surgery and postoperative care….”

[Training](https://rci.ucmerced.edu/iacuc/researchers/training-animal-researchers) is a legal requirement (AWAR §2.32), a Guide ‘must’. The UCM IACUC recognizes that individuals may have a wide range of educational backgrounds and experience that may require various levels of training before working with animals, including species-specific and aseptic surgery training. Personnel trained to perform human surgery may require additional instruction for interspecies variations in anatomy, physiology, and response to anesthetics and analgesics.

Regardless of an individual’s responsibility or educational background, all personnel performing surgery must have thorough knowledge and understanding of the IACUC-approved protocol procedures (working within the protocol’s limitations set forth by the PI and approved by the IACUC) and possess knowledge and familiarity with aseptic technique and the relevant aspects of species studied. Training generally consists of two parts, a general, aseptic technique class followed by a supervised research paradigm-specific training component provided by PI.

Veterinary Consultation: The UCM veterinarians are available for consultation to assist researchers in developing surgical protocols or protocols including, anesthesia or sedation, as outlined in AWAR §2.33.

The UC Merced IACUC Protocol (among other things) provides a written narrative of the surgical procedures. The narrative is designed to provide IACUC members with a concise, comprehensive, step-by-step description of the surgical procedure(s) from skin preparation to wound closure using lay language. The narrative will be evaluated by external organizations, such as AAALAC—International, during their campus site visits. The narrative description’s author is expected to address various items, including:

* Survival and non-survival surgeries
* Presurgical animal preparation
* Anesthetic induction, maintenance, and recovery
* Neuromuscular blocking agent use
* Intraoperative animal assessment
* Surgical narrative description
* Surgical procedure duration
* Methods to address:
* Hypothermia
* Dehydration
* Surgical complications
* Postoperative monitoring/complications

**II. Purpose:**

This document delineates the responsibilities and obligations of the Principal Investigator and study personnel performing survival surgical procedures.

**III. Definition:**

Aseptic Technique reduces microbial contamination to the lowest possible practical level and includes animal surgical preparation, such as hair removal and operative site disinfection; surgeon preparation, such as the provision of decontaminated surgical attire, surgical scrub, and sterile surgical gloves; sterilization of instruments, supplies, and implanted materials; and operative techniques used to reduce the likelihood of infection.

Survival Surgery is any surgical procedure where an animal regains consciousness for any period.

A Major Survival Surgery is any survival surgical procedure penetrating and exposing a body cavity or producing substantial impairment of physical or physiologic functions. Examples include laparotomy, thoracotomy, craniotomy, joint replacement, spinal transection, and limb amputation.

Multiple Survival Surgical procedures are not permitted on animals unless scientifically justified and IACUC-approved.

**IV. General Guidelines**

# Scheduling Surgical Procedures: In general, all survival surgical procedures are expected to occur Monday through Thursday of a five-day work week, not on Friday. Most complications occur within the first 24 hours and adhering to this schedule optimizes animal welfare, care, and scientific outcomes.

Procedures are expected to occur when animals can be continuously observed from anesthetic induction to the point they are sternal and making purposeful movements. More invasive procedures require more intense monitoring, including animal observations after hours or weekends. Sufficient personnel must be sourced and trained to meet these observation needs.

# Surgical Locations

A rodent and bird surgical area can be a room or portion of a room that is easily sanitized and not used for any other purpose during the time of surgery. However, because dedicated surgical facilities provide advantages, such as reduced investigator regulatory burden, consideration must be given for using dedicated UCM surgical facilities, whenever possible. There is no charge for the facilities or the gas anesthetic machines (when used within the facilities). An investigator’s laboratory may be used as a survival surgery area provided the investigator provides scientific rationale for such use, and the location is approved and undergoes a semiannual IACUC inspection. The facility must be available for regulatory (e.g., USDA) and voluntary accreditation organizations to inspect the area, which may occur without notice. In selecting a surgical location, investigators must bear in mind that “The number of personnel and their level of activity have been shown to be directly related to the level of bacterial contamination and the incidence of postoperative wound infection…” (the Guide, p. 144). Thus, every attempt should be made to sufficiently and physically separate the surgical area from other room areas to minimize unnecessary traffic and decrease potential wound contamination. Furthermore, the location should include three areas:

* A designated animal preparation area, including weighing, hair or feather removal, and initial skin disinfection. The preparation area should be sufficiently separate from the surgery area to minimize the potential for surgical area contamination by aerosols generated during animal preparation. Again, physical separation of the surgical preparation and surgical areas is expected.
* A separate surgical procedures area (i.e., from skin incision to wound closure). The surgical table and immediate surrounding areas must be sanitizable and cleaned (washed with soap and water) and then disinfected using appropriate agents (see Table 1 below). The immediate surgical area should be disinfected before and between surgeries to decrease dust-borne contamination and may not be used for other purposes during surgery.
* Finally, a separate recovery area should be established. This area should be a quiet, undisturbed location permitting animal observation. Properly managed, the animal preparation area may be suitable.

# Surgical Instruments

Surgical instruments must be sterile. Table 2, below, provides a list of acceptable instrument sterilization methods. Autoclaving is the preferred method of sterilizing instruments.

Surgical instruments may be used on more than one rodent; however, any item used on multiple animals must be autoclaved first, then carefully cleaned and disinfected between animals (see Table 3 below); start with autoclaved instruments, as autoclaving is the gold standard. Hot bead sterilizers are the preferred method to reuse instruments that were previously autoclaved, although prolonged soaking in disinfectant is also acceptable. Because disinfection effectiveness is directly dependent upon the contact time with the disinfectant, the surgeon is expected to anticipate the number of surgical instruments required to guarantee uninterrupted conduct of the procedures while affording ample disinfectant contact time. Replace disinfectants when contaminated with body fluids or tissues, to ensure disinfectant effectiveness. Insufficiently rinsed disinfectants on the instruments may cause skin or wound irritation.

| Table 1. Recommended Hard Surface Disinfectants\* | | |
| --- | --- | --- |
| Agents | Examples | Comments |
| Alcohols | 70% alcohol | Minimum contact time required is 15 minutes. Contaminated surfaces take longer to disinfect. Remove gross contamination before using. Inexpensive. Flammable. Alcohol is neither a sterilant nor a high-level disinfectant. |
| Chlorine | Sodium hypochlorite (Clorox® 10% solution)  Chlorine dioxide (Clidox®, Alcide®) | Corrosive. Presence of organic matter reduces activity. Chlorine dioxide must be fresh (<14 days old). Kills vegetative organisms within 3 minutes of contact. |
| Aldehydes | Glutaraldehyde Cidex®, Cide Wipes ®) | Rapidly disinfects surfaces. Toxic. Exposure limits have been set by OSHA. |

\*Table tops, equipment. Always follow manufacturers’ instructions.

Monitoring Sterility

Sterility indicators comprise chemical and biologic indicators. Chemical indicators frequently will have a color change when a given environmental condition develops, such as contact with a suitable temperature or contact with a sterilizing agent; this may not indicate the item(s) are sterile. Biologic indicators provide a higher level of assurance as they assess bacterial growth following indicator exposure to a given temperature and/or sterilizing agent (e.g., steam, ethylene oxide) for a given period. A sterility assessment program employing both chemical and biologic indicators is optimal.

Include sterility indicators inside the pack or pouch containing the items being sterilized, and an external indicator such as autoclave tape. These commercially available strips change colors to indicate the pack’s internal area has reached sterilization conditions. Write the sterilization date on the pack’s or pouch’s exterior, frequently on the external indicator (autoclave) tape.

Autoclaves must undergo a quality control assessment using biologic indicators monitored at least quarterly; this involves placing a biologic indicator within a pack. The tubes are removed after the sterilization process and incubated to assess the sterilization process. An autoclave log should be maintained including the dates instruments were sterilized (e.g., autoclaved) with the internal/external pack sterilization indicator. Likewise, the log should include the quarterly biologic indicator results. If either the chemical or biologic indicators fail, describe the actions taken to ensure the autoclave has returned to proper working order.

| Table 2. Recommended Methods of Instrument Sterilization\* | | |
| --- | --- | --- |
| Agents | Examples | Comments |
| \*\*\*Physical\*\*\* |  |  |
| Steam Sterilization | Autoclave | Effectiveness dependent upon temperature, pressure and time (e.g., 121°C for 15 min. vs. 131°C for 3 min.). Requires chemical and biologic indicators. |
| Dry Heat | Hot bead sterilizer | Fast (15 to 20 seconds). Instruments must be cooled before contacting tissue. Instruments must be sterilized by one of the other methods prior to the instruments’ first use that day. |
| \*\*\*Chemical\*\*\* |  |  |
| Gas Sterilization | Ethylene Oxide | Requires 30% or greater relative humidity for effectiveness against spores. Gas is irritating to tissue; all materials require safe airing time. Carcinogenic. Suitable for catheters and implants. |
| Chlorine Dioxide | Clidox®, Alcide® | A minimum of 6 hours required for sterilization. Corrosive. Presence of organic matter reduces activity. Must be freshly prepared (<14 days). Must be thoroughly rinsed from instruments using sterile distilled water before use. |
| Aldehydes | Glutaraldehyde | Many hours required for sterilization. Corrosive and irritating. Consult Biosafety Officer on proper use. Must be thoroughly rinsed from instruments using sterile distilled water before use. |

\* Always follow manufacturers’ instructions.

# Pre-Surgical Evaluation & Treatment

Pre-existing animal health conditions may negatively affect the immediate or long-term success of the surgical procedures and the research results’ fidelity. Pre-surgical animal evaluations help ensure the animals are not overtly ill; this includes a visual inspection of the animal and assessing their behavioral status. The animal should be bright, responsive, alert, well-hydrated and behaving normally and should have a smooth coat and clear eyes. Bring animals with physical or behavioral abnormalities to the attention of the UCM veterinary staff.

Withholding food or water in rodents or birds is generally unnecessary unless specifically mandated by the protocol or surgical procedure (e.g., gastrointestinal surgery). Withholding food or water for more than six hours should be discussed with a UCM veterinarian.

In some cases, it may be preferable to initiate antibiotic or analgesic treatment before surgery. The UCM veterinarian is available to discuss and develop antibiotic or analgesic treatment regimens.

| Table 3. Recommended Instrument Disinfectants\* | | |
| --- | --- | --- |
| Agents | Examples | Comments |
| Steam Sterilization | Autoclave | Effectiveness dependent upon temperature, pressure and time (e.g., 121°C for 15 min. vs. 131°C for 3 min.). Requires chemical and biologic indicators. |
| Dry Heat | Hot bead sterilizer | Fast acting. Instruments must be cooled before contacting tissue |
| Alcohols | 70% alcohol | Minimum contact time required is 15 minutes. Contaminated surfaces take longer to disinfect. Remove gross contamination before using. Inexpensive. Flammable.  Alcohol is neither a sterilant nor a high level disinfectant. |
| Chlorine | Sodium hypochlorite (Clorox® 10% solution)  Chlorine dioxide  (Clidox®, Alcide®) | Corrosive. Presence of organic matter reduces activity. Chlorine dioxide must be fresh (<14 days old). Kills vegetative organisms within 3 min. Must be thoroughly rinsed from instruments using sterile distilled water before use. |
| Peracetic Acid Hydrogen Peroxide | Spor-Klenz® | Corrosive to instrument surfaces. Must be thoroughly rinsed from instruments using sterile distilled water before use. |
| Aldehydes | Glutaraldehyde | Minimum contact time required is 15 min. Corrosive and irritating. Consult Biosafety Officer on proper use. Must be thoroughly rinsed from instruments using sterile distilled water before use. |

\* Always follow manufacturers’ instructions.

# Surgical Preparation

## Hypothermia

Heat loss is a significant concern for rodents and birds undergoing anesthesia. Heat loss occurs rapidly and affects various physiologic parameters and prolongs an animal’s time to first purposeful movement and restoration of the righting reflex. Hypothermia may have additional downstream deleterious research effects, too. Generally, under standard husbandry conditions, rodents are maintained at a temperature below their thermoneutral zone. Anesthetic use exacerbates hypothermia by disrupting various mechanisms that maintain a conscious animal’s body temperature. As the anesthetic agent and its hypothermia inducing effects wane during anesthetic emergence, physiologic responses such as vasoconstriction may contribute to heat loss by reducing the heat absorbed from external heat sources. Additional contributors include skin preparation (e.g., surgical scrub, alcohol) for surgical procedures. Hypothermia will yield a deepened anesthetic plane and can confound research outcomes in anesthetized animals, not undergoing surgery (e.g., restraint), used in studies including hepatic, cardiac, brain, and urogenital systems.

Heated water recirculating heating pad systems are preferred and recommended as they virtually eliminate the likelihood of burns. Electric heating pads may overheat, unevenly heat, or burn the animal; if used, the pad must be set on low, and insulating layers (e.g., light cloth covering, bubble wrap) placed between the animal and the heating pad. The animal must be observed frequently for signs of hyperthermia. Electric heating pads are associated with a high incidence of patient burns; likewise, systems using a rectal probe rheostat can overheat and burn animals if the rectal probe either falls out or not inserted far enough into the rectum (see manufacturer’s recommendations). Because heat lamps may cause severe hyperthermia or other thermal injuries, their use is prohibited.

The SQ or IP administration of warm fluids (5-10 ml/kg) is highly recommended at the end of an anesthetic procedure or intermittently during prolonged procedures. This serves to replace lost water through insensible loss (e.g., respiration), correct dehydration of exposed body cavities, and facilitate the excretion/correct water losses associated with some injectable anesthetics (e.g., ketamine, xylazine). Administering fluids such as lactated Ringer’s solution is preferred, but normal saline is acceptable.

## Animal Preparation

Animal surgical preparation includes removing hair/feathers from the surgical site with a generous border (at least 1 cm) to avoid incision site contamination. Perform the hair/feather removal in a location separate from the surgical area while keeping the animal warm by a heating pad or placing an insulating layer (e.g., towel, bubble wrap) between the animal and an unheated surface. When using clippers to remove the fur, wetting the fur prior will minimize personnel allergen exposure. When removing the hair/feathers with a vacuum cleaner, the vacuum cleaner must be HEPA filtered. Scrub the surgical site with a germicidal scrub (e.g., Betadine®), being careful to scrub from the center of the surgical site towards the periphery in a circular manner. Rinse the site with 70% alcohol in a similar manner. Avoid using excessive surgical scrub/alcohol to minimize their cooling effects on the patient. At least three alternating preparations of germicidal scrub and rinse are considered adequate, preferably followed by spraying the area with germicidal solution. Finally, drape the area with sterile drapes to prevent contaminants from entering the surgical field and provide a sterile area to place instruments and sterile items during surgery. Researchers may wish to consider using cling film, such as Press’n Seal®, to drape the surgical site.

## Surgeon Preparation

Surgeons must thoroughly wash their hands with a bactericidal scrub. The use of sterile surgical gloves is required. Gloves dipped in bleach are unacceptable. A surgical mask must be worn for major surgeries and implanted devices but is highly recommended for minor procedures. Wearing a clean lab coat is mandatory; however, a sterile gown is preferable, especially for major surgeries or surgeries involving implanted materials.

# Anesthesia

UCM veterinarians can assist in selecting anesthetic regimens as researchers develop their IACUC approved research protocol. Generally, gas anesthesia (e.g., isoflurane) is highly recommended because it is generally safer and more easily controlled than injectable agents, plus it provides oxygen as a carrier gas. In any case, the animal must be fully anesthetized before initiating the procedure and maintained in a consistent anesthetic plane throughout the surgery. Anesthetic depth may be monitored in several ways (e.g., respiration rate, corneal reflex, positive toe pinch) and may vary depending upon the species and the anesthetic agent used. For rodents and birds, it is generally not feasible to monitor the heart rate, unless suitable electronic instrumentation is available.

For guidance in selection and use of anesthetics, please contact a UCM veterinarian.

# Surgical Procedures

All aspects of the surgical procedures must be conducted as described in the IACUC approved protocol. Animal monitoring during surgery is critical. In addition to monitoring anesthetic depth as described above, maintaining normal body temperature is of particular importance, as anesthetics can directly or indirectly induce hypothermia.

To prevent corneal desiccation, place bland ophthalmic ointment in both eyes immediately following anesthetic induction. If the animals are undergoing survival stereotaxic surgery, blunt ear bars must be used to prevent tympanic membrane damage.

Paralytic agents must not be used without sufficient anesthesia and appropriate monitoring. Paralytic agents must be described in the IACUC protocol along with monitoring methods.

# Suture Selection

Use an absorbable suture material for body wall closure or other internal wound closures, and a nonabsorbable monofilament suture material for skin closure. Although slightly more technically challenging, subcuticular suture placement is an acceptable skin closure using absorbable materials. The smallest gauge suture material should be used as practicable; typically, 3-0 or 4-0 material is acceptable. A list of acceptable suture materials is included in Table 4 below.

| Table 4. Recommended Suture Materials | |
| --- | --- |
| Suture Material | Characteristics and Frequent Uses |
| Vicryl®, Dexon® | Absorbable; 60-90 days. Suitable for internal wound closure. |
| PDS®, Maxon® | Absorbable; 6 months. Suitable for internal wound closure where extended wound support is desirable. |
| Prolene® | Nonabsorbable. Suitable for skin closure. |
| Nylon | Nonabsorbable. Suitable for skin closure. |
| Stainless Steel Wound Clips, Staples | Nonabsorbable. Suitable for skin closure. Requires instrument for removal from skin. |

Because silk and chromic gut may cause tissue inflammation, these materials are not acceptable for wound closure.

When presented with a choice of braided or monofilament suture material, researchers are strongly advised to use monofilament material. Bacteria and biofilm infiltration into suture material is well known, and braided material is categorically more problematic than monofilament material. There is a subtype of monofilament termed ‘barbed,’ and this material should be avoided whenever possible as it has been demonstrated to harbor bacteria more than conventional monofilament materials.

Sutures, staples, or wound clips must be removed 7-14 days following surgery. Suture removal before euthanasia is not necessary for those animals euthanized within 14 days of surgery. Foreign substances (e.g., suture material) remaining in the incision for an extended period serve as a nidus of irritation and infection. Please contact a UCM veterinarian to examine any incisions that do not appear to be healing.

For additional guidance with suture selection and use, please contact a UCM veterinarian.

# Post-operative Recovery

Researchers must observe animals during post-surgical recovery. The animal, whether recovering in or out of its ‘home’ cage, must be kept warm. Animals administered anesthetic or analgesic agents will not have the same responses to painful stimuli as conscious counterparts; therefore, water-circulating heating pads are recommended. Electric heating pads may overheat or burn the animal; if these are used, the pad must be set on low, and a light cloth covering should be placed between the animal and the pad, and the animal must be observed frequently for signs of hyperthermia. Turn somnolent animals periodically to prevent burns or thermal injury. Provisions must also be made for a conscious animals to escape the heat source when it becomes too warm, such as placing the cage over only half of the heat source. Because heat lamps may cause severe hyperthermia or other thermal injury, their use is prohibited.

Continuously watch recovering animals until in sternal recumbency and showing a degree of purposeful movement. Animals emerging from anesthesia commonly slip back to a more somnolent or ‘sleepy’ state. Unconscious animals must never be unattended. To prevent undue injury of co-housed animals, individually house rodents until fully recovered from the anesthetic.

Warm lactate Ringer’s solution (LRS) fluids are also important as animals undergoing routine anesthetic procedures are prone to developing dehydration, especially those using injectable anesthetic agents such as xylazine or dexmedetomidine, which cause elevated blood glucose levels leading to increased urine production.

Investigators are kindly asked to identify the cages of post-operative animals and/or animals which have undergone anesthesia.

# Post-operative Analgesic and Antibiotic Treatment

Analgesia

* Analgesia must be provided to all animals following survival surgery unless scientific justification for withholding post-operative analgesics is provided by the investigator and approved by the IACUC, or if a veterinarian examines the animal and determines analgesic administration is no longer necessary.  In cases where post-operative analgesics cannot be administered for scientific reasons, the animals must be listed as Pain Category E (i.e., pain/distress cannot be relieved by use of anesthetics, analgesics, or tranquilizers as the use of these agents would interfere with the experimental design).
* Analgesia and animal welfare benefits from a multimodal analgesia strategy. Multimodal analgesia combines various analgesic classes (e.g., opiates, NSAIDs, local anesthetics) to optimize pain management, which confers significant advantages over a single analgesic agent.
* Local anesthetics such as Marcaine® (bupivacaine) or lidocaine, in addition to systemic analgesia, may be indicated for some procedures resulting in significant skin disruption (e.g., Alzet pump placement, catheter exteriorization), as agents block the pain cascade’s onset resulting from dermal nerve cell disruption. Due to their short duration, local analgesics are not intended for use in lieu of systemic analgesics, unless withholding systemic analgesia is scientifically justified in the IACUC-approved protocol. Local anesthetics are typically administered before the animal’s anesthetic emergence. Local anesthetic blocks are a strongly recommended multimodal analgesia component.
* Administer analgesia so that it is effective prior to the animal’s emergence from anesthesia. The use of preemptive analgesia is expected; researchers are reminded that using preemptive analgesia may impact anesthetic depth and, therefore, gas anesthesia is preferred over injectable due to its greater anesthetic control.
* When injectable anesthesia (e.g., ketamine/dexmedetomidine) is used with a reversal agent (e.g., atipamezole), researchers are reminded that these anesthetic reversal agents reverse the agent providing the analgesia and, therefore, another postoperative analgesic agent must be administered prior to the reversal agent. Some analgesics, such as buprenorphine, may require 30-45 minutes to take effect.
* Major survival surgeries require at least 72 hours of post-operative analgesia, unless otherwise indicated in the professional judgement of the veterinarian. For major survival surgeries, consultation with the veterinarian should also include consideration of pre-operative analgesia. Again, a major survival surgery is defined as any procedure that penetrates and exposes a body cavity, produces substantial impairment of physical or physiologic functions, or any procedure that requires the use of more than a single application of a short-term anesthetic.
* Minor procedures require at least 24 hours of post-operative analgesia, unless otherwise indicated in the professional judgement of the veterinarian.
* For guidance on selection and use of anesthetics, analgesics, and/or tranquilizers please contact a UCM veterinarian.
* There are several long acting opiate (buprenorphine) compounds available on the market that provide analgesia for 2-3 days following a single administration. While these compounds are highly recommended, animals must be assessed at least daily during the postoperative period.
* A list of commonly used analgesics is included in Table 5 below.

Antibiotics

Discuss postoperative antibiotic therapy with a UCM veterinarian to determine if routine antibiotic administration is necessary. Surgical procedures performed with strict adherence to aseptic technique may not require antibiotics. Additionally, postoperative antibiotics should be provided if the animal will survive long enough to develop an infection, but may depend upon other factors such as the procedure’s invasiveness, the animal’s immune status, etc. Preoperative antibiotic administration can further minimize postoperative infections.

Administer antibiotics parenterally to the animal as it has been shown that antibiotics administered in the drinking water and diet yield subtherapeutic antibiotic levels. The administration should occur SQ or IP; additionally, diluting antibiotics such as Baytril® (enrofloxacin) in sterile normal saline is recommended to avoid injection site skin reactions that may develop following administration of the undiluted compound.

# Long-Term Recovery and Monitoring

Post-surgical observations include a minimum daily observation, including weekends and holidays, of the animal’s condition and the surgical site. Animals should be observed for continued recovery, including state of arousal, indices of pain or discomfort, condition of the surgical wound, appetite, hydration status, capillary refill time, mucous membrane color, or fecal and urine production.

Some surgical manipulations may require an extended postoperative monitoring period. In consultation with the investigator, the UCM veterinary staff can determine the appropriate duration and extent of monitoring. Some situations constituting prolonged monitoring periods include animals with chronic debilitating disease states (e.g., diabetes mellitus), animals undergoing organ transplantation or immunosuppressive therapy, and animals with chronically implanted instruments or catheters.

Contact the veterinarian in the event that post-surgical complications are observed, including after hours, weekends, and holidays.

| Table 5. Commonly Used Analgesics and Dosing Information | | | | | |
| --- | --- | --- | --- | --- | --- |
| Class | Drug | Species | Dose Range  (Recommended) | Route\* | Frequency |
| Opiates\*\* | Buprenorphine | Mouse | 0.5-1 mg/kg (0.75) | SQ | 6-8 hours |
| Rat | 0.01-0.05 mg/kg (0.025) |
| Buprenorphine SR | Mouse | 0.3-1 mg/kg (0.5) | SQ | Every 3 days |
| Rat | Rat: 0.3-1.2 mg/kg (0.6) |
| Buprenorphine XR  Trade Name: ETHIQA | Mouse | 3.25 mg/kg | SQ | Every 2-3 days |
| Rat | 0.65 mg/kg |
| NSAIDs | Carprofen | Mouse | 5-10 mg/kg (7) | SQ | Daily |
| Rat |
| Ketoprofen | Mouse | 5 mg/kg | SQ | Daily |
| Rat |
| Flunixin meglumine | Mouse | 1.1-2.5 mg/kg (1.8) | SQ | 12 hours |
| Rat |
| Local  Anesthetics | Lidocaine | Rat/Mouse | 2-4 mg/kg | SQ | — |
| Bupivacaine | Rat/Mouse | 1-2 mg/kg | SQ | — |

\*SQ: Subcutaneous; \*\*Opiates are controlled substances with record keeping and other requirements

# V. Record-keeping Requirements

As per [AAALAC—International](https://www.aaalac.org/accreditation-program/faqs/#D4) recommendations, surgical records are expected for rodents and birds. These records should include the administration of anesthetics, fluids, and any drugs; procedure details, including intraoperative monitoring; daily post-operative recovery observations and treatment, including administration of analgesics and antibiotics; monitoring of incision healing, including suture/staple removal if applicable; and the initials of the individual performing these tasks. Record all medications, including the name, dose, route, and time of administration. Additionally, any adverse outcomes should be noted. A sample Procedure Record for Rats, Mice, and Birds and a Post-operative Evaluation Record for Mice, Rats, and Birds are available on the IACUC website. Please complete the anesthetic record portion for procedures lasting more than 1 hour.

To facilitate the veterinary staff’s evaluation of post-operative healing and to ensure sutures are appropriately removed, each cage card must be clearly marked with the date of surgery.

All records relating to surgical procedures and post-operative care may be subject to review during IACUC inspection, Post-approval Monitoring (PAM), AAALAC site visit, and/or USDA inspection. These records must be available; ideally, the original records should be available and maintained centrally so ready access is available when these reviews occur.

Wound Management

The images below are designed to provide researchers guidance on assessing surgical wounds.

| Appearance | Score | Action/Characteristic Images | | |
| --- | --- | --- | --- | --- |
| Clean, Dry, Intact Surgical Site | 0 | No action required | | |
| Image | Image | Image |
| Open, reddened, and/or mild surgical site discharge ≤ 24h | 1 | Anesthetize the animal and close an open wound. Consult with veterinarian or veterinary technician. | | |
| Image | Image | Image |
| Open, reddened, and/or mild surgical site discharge ≥ 24h | 2 | Euthanize the animal or contact the veterinarian for wound management advice. | | |
| Image | Image |  |
| Open, reddened, and/or mild surgical site discharge ± yellow/white crusty, dried material. | 3 | Euthanize the animal or contact the veterinarian for wound management advice. | | |
| Image | Image |  |

# VI. References

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