POLICY ON ASEPTIC RECOVERY SURGERY ON USDA REGULATED NONRODENT SPECIES

Adopted by the University Committee on Animal Resources
October 15, 2014

The U.S.D.A Animal Welfare Act (9 CFR) requires use of aseptic technique when performing major and minor invasive recovery surgery on USDA-regulated species. Major invasive surgery includes penetration and exposure of the cranium, thorax, abdomen or any procedure producing permanent impairment of physical or physiological functions. Minor invasive surgery does not penetrate a body cavity and includes relatively minor operative procedures such as blood vessel cut down, corneal surgery and eye coil implantation. As required by the U.S. Public Health Service and the University Committee on Animal Resources (UCAR), all vertebrate animal-use protocols, regardless of the funding source, must comply with the guidelines stated in the Guide and the requirements of the USDA Animal Welfare Act.

This policy refers to USDA regulated nonrodent species. If you are working with any rodents covered by USDA regulations such as hamsters, gerbils, mole rats or prairie voles, please refer to the Policy on Aseptic Recovery Surgery on Rodents and Birds.
Investigators who believe that their nonrodent USDA regulated animals require exceptions to the Guide recommendations or USDA requirements should contact UCAR for assistance. Otherwise, investigators using these species are expected to follow this policy.

MAJOR INVASIVE SURGERY

Pre-Operative Animal Preparation

All animals must be fasted 4 to 20 hours prior to general anesthesia to empty the stomach of ingesta. Free choice water is made available with the exception of water scheduled animals which may over consume. Because they cannot vomit, rabbits do not require fasting unless there is a need to empty the stomach for an abdominal surgical procedure. Under most circumstances, animals receive the first dose of an anesthetic drug within their home cages using a squeeze cage for macaques or manual restrained for animals which are safe to handle, such as new world primates, rabbits, dogs, cats, and farm animals.

Animal preparation must be performed in a dedicated, physically separated area from the Operating Room. Hair must be removed from the surgical site with clippers, a razor or a medical depilatory. The surgical site must be disinfected with the following two-step process repeated three times:

1. Gross contamination should be removed by using a surgical scrub at the surgical site (chlorhexidine or povidone iodine scrub) using appropriately sized gauze sponges starting from the center of the shaved surgical site moving outward.

2. The surgical site should then be wiped povidone iodine or chlorhexidine solution using appropriately sized gauze sponges starting from the center of the surgical site moving outward.

Operating Room & Aseptic Technique
Surgery must be conducted in an Operating Room (O.R.) physically separated from the other functional zones (Animal Prep, Surgeon Prep, Instrument Prep and Recovery). Air pressure differential for the O.R. must be positive to all other adjacent spaces.
The temperature in the surgery room should be increased and/or the animal placed on a covered warming device (e.g. circulating warm water blanket) to prevent hypothermia.
A sterile drape is required over the surgical site to avoid sterile instruments, sterile gloves or exposed viscera from coming in contact with unprepped areas. The surgeon must wear scrubs, a sterile surgical gown, sterile gloves, shoe covers, a face mask and a head cover.
Monitoring of anesthesia must be documented using the ANESTHESIA LOG (www.urmc.rochester.edu/vivarium).

• **Submit completed anesthetic records to DLAM when animal is returned to housing in stable condition and can be left alone.**

A dedicated anesthetist should observe mucous membrane color, respiratory rate and pattern, body temperature and monitor for the loss of pedal, corneal and pinnal (external ear) reflexes. More sophisticated methods of patient monitoring including EKG, pulse oximetry, end tidal CO2, blood pressure measurements and blood gas measurements are highly recommended.

The surgeon must maintain aseptic technique by only touching sterile instruments or sterile surfaces. If the surgeon breaks aseptic technique by touching a nonsterile surface, he/she must don new sterile gloves.

The abdominal or thoracic body wall is to be closed with absorbable suture material. The skin should be closed with staples or with a nonabsorbable suture material in a simple interrupted pattern or absorbable sutures in a continuous subcuticular pattern. Absorbable sutures placed in a subcuticular pattern to close the skin need not be removed postoperatively since they are buried under the skin. All other skin sutures or staples should be removed seven to ten days after surgery. Investigators should consult with veterinary staff regarding appropriate closure techniques if not familiar with the models.

Instrument Preparation and Area
All instruments must be sterilized, but the method of choice may vary depending upon the surgical instruments or devices used. Acceptable sterilization techniques include autoclaving using steam under pressure, ethylene oxide (EO) or cold sterilization. Approved cold sterilization methods include: soaking instruments in 2.5 – 3.5% glutaraldehyde (e.g. Cidex Plus for 10 hrs. at 20-25° C) or 7.5% hydrogen peroxide (e.g. Sporox Sterilizing and Disinfection Solution for 6 hours at 20° C) according to manufacturer's instructions.

Monitoring of Autoclave Equipment
Heat sensitive chemical indicators must be used to verify that surgical instruments and other materials are appropriately sterilized. Investigators must use one autoclave integrator strip in each pack to be autoclaved. The strip should be placed in a location considered to be the hardest for the steam to reach. Place autoclave tape on the pack surface. Contact DLAM for more information about these methods.

Instruments may be cleaned, wrapped and sterilized in a room separate from the animal prep room and the O.R. or may share the surgeon prep room as long as a different sink is used for each function.

Surgeon Pre-Operative Preparation and Area
The surgeon pre-operative preparation area must be physically separated from the pre-operative animal preparation area and the operating room. The area may be shared with instrument preparation but separate sinks are required.

The surgeon must don a face mask, cap, shoe covers and surgical scrub top and bottom before scrubbing hands. The surgeon must wash his/her hands with an antiseptic surgical scrub for a minimum of three minutes using ten scrubs per surface working from the finger tips down and then aseptically put a sterile gown followed by sterile gloves.

Animal Recovery and Area
Recovery from a surgical plane of anesthesia may be staged with first steps occurring in the O.R. where physiological parameters (heart rate, PO2, respiratory rate, return of reflexes, ..) may be safely monitored. Final stages of recovery may occur in the animal room enclosure (e.g. primates) or in the animal preparation area in a recovery transport cart (e.g. dog, cat, swine) before being returned to the animal housing room.

Criteria for assessing when it is safe to remove the endotracheal tube include: an easily elicited tracheal cough, an increase in jaw tone and resumption of swallowing activity. Animals should be recovered from anesthesia in a warmed environment. Post procedural or anesthetized animals may not be left unattended until fully recovered, able to ambulate, with pink mucous membranes and stable respirations. Close observation provides the opportunity for early detection and response to potentially life-threatening problems.
The responsible individual must record the time the animal is returned to housing on the DLAM POST-OP RECORD (www.urmc.rochester.edu/vivarium). The person must also describe the animal's condition by recording the quality and/or rate of respirations, mucous membrane color and/or capillary refill time and the response of various reflexes (e.g. palpebral, corneal, righting reflexes) and quality of jaw tone. Pertinent intra-operative complications, post-operative orders or observations should be recorded on the Post-Op Chart. The individual writing post-operative orders must make sure that antibiotic and/or analgesic agents, dosages, routes and treatment intervals are included on the chart. Investigators must designate who is responsible for providing post-op medication (DLAM or PI's Staff). Post-op orders must be the same as those stated in the UCAR protocol or as directed by a veterinarian. The Post-Op Chart must be delivered to Animal Resource office (G6708) during working hours or the DLAM Completed Forms mailbox after business hours.

The DLAM veterinary staff routinely monitors all post-op USDA regulated nonrodent species for a minimum of three days after surgery. During this time, the investigator will be informed of any complications observed.

MINOR INVASIVE RECOVERY SURGERY

Minor invasive surgery does not penetrate a body cavity and includes relatively minor operative procedures such as Lasik™ corneal surgery and eye coil removal.
Pre-operative animal and surgeon preparation and intra-operative procedures for minor invasive surgery on regulated species does not require a dedicated room. Surgeons must wear sterile gloves, mask and use sterile surgical instruments. Animal preparation techniques, aseptic procedures, anesthetic depth monitoring, recovery methods and the associated documentation must be followed as described for major invasive surgery above.

Anesthetics and Analgesics

Anesthetics and analgesics must be administered as described in the UCAR approved protocol. Systemic analgesics should be administered to all species experiencing major survival surgical procedures for a minimum of three days following surgery. Animals undergoing minor procedures that may result in post-op discomfort must also receive analgesics. Analgesics administered prior to the surgical manipulation are beneficial for pain relief in laboratory animals; therefore pre-emptive analgesic therapy is required. Drugs must be given at the dosing interval stated in the UCAR protocol. The decision to discontinue analgesic therapy should be made based on the observation that the animal appears to be comfortable at the end of the previous dosing interval (i.e. when the next analgesic treatment is due).

The following formulary contains standard drugs used and recommended by DLAM veterinary staff. This formulary may be adjusted as new drugs are discovered or new research indicates more effective and/or safer analgesic drugs in these species. Investigators should consult with a veterinarian when planning a protocol for the most appropriate anesthetic and analgesic regimen specific to that surgical procedure and research use.

Anesthetics and Analgesics used in Ferrets

Anesthesia in Ferrets	**Dose & Route**
Ketamine + Xylazine	25 mg/kg + 2.5 mg/kg IM
Isoflurane	0 - 5 % to effect
Analgesia in Ferrets	
Buprenorphine (Buprenex)	0.01 - 0.03 mg/kg SQ, IM or IV every 8 - 12 hours
Flunixin (Banamine)	0.5 – 2.0 mg/kg SQ, IV 12 - 24 hours

Anesthetics and Analgesics used in Rabbits

Anesthesia in Rabbits	**Dose & Route / Comments**
Ketamine + Xylazine	44 mg/kg + 5 mg/kg SQ* or IM. SQ is preferred route of administration Xylazine can be reversed with 0.2mg/kg

	yohimbine SQ or IV.
Ketamine + Dexmedetomidine	15-25mg/kg + 0.05-0.1mg/kg SQ Dexmedetomidine can be reversed with 0.2mg/kg atipamezole SQ or IV.
Acepromazine	0.25 – 0.75 mg/kg IM for blood collection from central ear artery
Isoflurane	1 - 3 % to effect
Analgesia in Rabbits	
Buprenorphine (Buprenex)	0.01 - 0.05 mg/kg SQ every 6 -12 hours
Flunixin (Banamine)	1 - 2 mg/kg SQ every 12 - 24 hours
Meloxicam (Metacam)	0.2 mg/kg SQ or 0.3 mg/kg PO once a day

Anesthetics and Analgesics used in Cats

Anesthesia in Cats	**Dose & Route / Comments**
Ketamine + diazepam	10 mg/kg + 0.5 mg/kg IV (mix together). Give 50% dose, then give smaller volumes as needed for induction
Ketamine + Dexmedetomidine	5mg/kg + 0.04mg/kg IM
Isoflurane	1 - 3 % to effect
Analgesia in Cats	
Buprenorphine (Buprenex)	0.004 - 0.01 mg/kg SQ every 8 -12 hours
Sustained Release Buprenorphine	0.12mg/kg SQ every 72 hours
Meloxicam (Metacam)	0.2 mg/kg PO, IV, SQ on Day 1; then 0.1 mg/kg once a day subsequent days

Anesthetics and Analgesics used in Dogs

Anesthesia in Dogs	**Dose & Route / Comments**
Ketamine + diazepam	10 mg/kg + 0.5 mg/kg IV – *mix together and give 50% dose, then in small increments as needed – for induction*
Ketamine and Dexmedetomidine	2mg/kg + 0.01mg/kg IM
Isoflurane	1 - 3 % to effect
Propofol	4-6mg/kg IV, slowly to effect May cause apnea with rapid administration
Analgesia in Dogs	
Buprenorphine (Buprenex)	0.01 - 0.04 mg/kg SQ every 8 -12 hours

Sustained-Release Buprenorphine	0.03-0.06mg/kg SQ every 72 hours
Meloxicam (Metacam)	0.2 mg/kg PO, IV, SQ on Day 1; then 0.1 mg/kg once a day for subsequent days

Anesthetics and Analgesics used in NHP

Anesthesia in the NHP	**Dose & Route / Comments**
Sodium Pentobarbital (25 mg/kg)	IV calculated dose given to effect, Atropine (0.04 mg/kg) IM or IV prevents bradycardia. Only recommended for perfusions.
Ketamine + diazepam	10 - 15 mg/kg + 0.25 - 0.5 mg/kg IM for CHEMICAL RESTRAINT ONLY FOR NONINVASIVE PROCEDURES or FOR INDUCTION Midazolam is preferred over diazepam for IM injection because midazolam is tissue soluble
Ketamine + Midazolam	10-15mg/kg + 0.25mg/kg IM Chemical restraint only for noninvasive procedures or as a premedication
Isoflurane	1 – 3 % to effect
Propofol	2-4mg/kg IV, slowly to effect For induction of anesthesia following pre-medication as an alternative to face masking
Analgesia in the NHP	
Meloxicam (Metacam)	0.1 – 0.2 mg/kg IM once a day (0.2 mg/kg on day one, then 0.1 mg/kg)
Meloxicam SR	0.6 mg/kg SQ
Flunixin	1.1 mg/kg IM, SQ every 12 - 24 hours
Buprenorphine (Buprenex)	0.01 – 0.04 mg/kg SQ every 6 -12 hours If the lowest dose (0.01mg/kg) is chosen, it must be given every 6-8hrs. Higher doses (0.03mg/kg) may be administered every 12hrs. (Nunamaker 2013).
Buprenorphine SR	0.06mg/kg SQ every 72 hours

Anesthetics and Analgesics used in Pigs

Anesthesia in Pigs	**Dose & Route**
Ketamine + Acepromazine	33 mg/kg + 1.1 mg/kg SQ
Isoflurane	1 – 3 % to effect
Propofol	2-4mg/kg IV, slowly to effect
Analgesia in Pigs	
Buprenorphine (Buprenex)	0.01 - 0.05 mg/kg IV or SQ every 6 -12 hours
Flunixin meglumine (Banamine)	0.5 – 1.0 mg/kg SQ, IV every 12 - 24 hours. *Five day maximum treatment*
Carprofen	3-4mg/kg PO every 12 hours, or IM every 24 hours
Meloxicam	0.4mg/kg PO or SQ every 24 hours