A. PURPOSE:
1. This document was created by the ULAM veterinary staff as a guideline for anesthesia monitoring during surgery and sedation. This is not intended to be an inclusive tutorial on all possible methods and all equipment available for anesthesia monitoring.
2. Current veterinary anesthesia standards of care focus on reducing anesthetic morbidity. Adverse changes in normal animal physiology can be detected and corrected early through responsible use of anesthesia monitoring equipment and trained personnel dedicated to anesthesia monitoring. For more detailed information regarding monitoring capabilities and physiological systems, please reference:
   a) AAHA Anesthesia Guidelines for Dogs and Cats
   b) 2009 ACVA Small Animal Monitoring Guidelines
3. Detailed information regarding pre- and post-operative care for animals undergoing survival surgical procedures as well as requirements for species specific anesthesia and analgesia are located on the UCUCA website at http://www.ucuca.umich.edu/guidesurg.htm.
4. If you have questions or comments about this document, please contact the ULAM veterinary staff at ulam-vets@umich.edu or 734-936-1696. The ULAM training core (ulamtraining@umich.edu or 734-763-8039) can be contacted to provide training in these techniques at no charge.

B. DEFINITIONS:
1. Anesthesia: Temporarily induces loss of sensation with or without loss of consciousness. Does **NOT** provide pain relief.
3. Sedation: A mild degree of central depression in which the patient is awake but calm.
   a) *Sedation is not sufficient for surgical procedures.*

C. RESPONSIBILITY:
1. It is the responsibility of the Principal Investigator to ensure appropriate anesthesia, monitoring, and analgesia is provided for all animals undergoing surgical or sedation procedures.

D. PROCEDURES:
1. Recovery Surgical Anesthesia Expectations:
   a) Procedures must meet current veterinary anesthesia standards of care.
b) Principle Investigators (PIs) are expected to purchase anesthesia and monitoring equipment corresponding to the complexity and potential anesthetic complications associated with their surgical research model.

c) Lab personnel are expected to maintain aseptic technique while providing appropriate anesthetic monitoring. Typically, this requires one person dedicated to surgery and a separate person dedicated to anesthetics monitoring.

d) Use of room air as a carrier for anesthetic inhalants is **highly discouraged**. If not using oxygen for a carrier, a pulse oximeter should be used to monitor blood oxygenation saturation throughout the surgical procedure.

2. Non-Rodent Recovery Surgical Anesthesia and Monitoring

   a) Pre-surgical examination: A physical examination should be performed including: heart rate and rhythm, mucous membrane color (normal = pink), capillary refill time (normal <2 seconds), respiratory sounds and rate, and temperature. It is recommended to perform the following baseline clinical chemistries: packed cell volume (PCV) or hematocrit, total protein, glucose, blood urea nitrogen (BUN), and serum creatinine.

   b) Monitoring: Monitoring must be performed throughout the anesthetic episode and until the animal is ambulatory. Monitoring frequency must be at a minimum of every 15 minutes. More frequent monitoring of every 5-10 minutes is highly recommended. Minimal monitoring requirements include:

   ➢ Cardiovascular System:

   (i) **Heart rate or pulse rate must be monitored**

   - Heart rate can be monitored by palpating the chest wall, placement of an esophageal stethoscope, or use of a standard stethoscope.
   - Pulse rate can be monitored by palpating a pulse on the extremities or through monitoring equipment such as a doppler ultrasound, pulse oximetry or direct arterial blood pressure.
   - An electrocardiogram can be used in conjunction with pulse oximetry or palpation to assess cardiac rhythm or detect cardiac dysrhythmias.

   (ii) **Peripheral perfusion must be assessed** by capillary refill time (normal < 2 seconds) or mucous membrane color (normal = pink).

   (iii) Blood pressure monitoring is highly recommended.

   ➢ Respiratory System:

   (i) **Respiratory rate must be monitored**.

   - Respiratory rate can be assessed by visually counting chest wall inflations, utilizing a stethoscope, monitoring rebreathing bag movements, or by appropriate monitoring equipment such as mechanical ventilation bellows or capnography.
   - For most species, tidal volume = 10-20 ml/kg with inspiratory pressures < 20 mm H2O.

   ➢ Body Temperature:

   (i) **Body temperature must be monitored during anesthesia and recovery** to prevent serious changes from normal. Thermal support should be supplemented depending on species, procedure, and duration of procedure.
• Warming devices should provide gentle heat only (maximum of 102°F). A thermometer can be placed next to the supplemental heat source to verify temperatures are < 102.

• **AVOID commercial human electric heating pads** – maximum temperatures are not well regulated and uneven surface temperature gradients exist risking thermal injury burns to the animal.

  When using a heating pad, an insulated pad or folded drape must be placed between the animal and the heat source.

• Heating lamps are discouraged since they are a fire hazard risk and the maximum temperature is difficult to regulate. However, heat lamps can be used as long as they are placed an appropriate distance away from the animal to ensure thermal burns do not occur.

  (ii) Recommended warming devices for non-rodents
  • Circulating water blanket (Gaymar®)
  • Forced air (Bair Hugger - Arizant®)
  • Snuggle Safe warmer and insulating pad
  • IV fluid warmers

➢ Anesthetic Depth:

  (i) **Adequate anesthetic depth must be evaluated during anesthetic period and can be assessed by the following:**

  • Loss of response to stimulation including toe, ear, or tail pinch
  • Loose mandibular jaw tone
  • Absent blink reflex
  • Eye position

    Non-ruminants – Eyes rotate rostroventrally

    Central eye position indicates depth too deep or too light

    Ruminants – Eyes rotate ventrally and only sclera is seen

    Central eye position indicates depth too deep or too light

  c) **Records:** Each surgical procedure must have a record of parameters monitored throughout the procedure. Each record should include the name of the surgery, the duration of the surgery, the anesthetic administered, the dose, and the values of all parameters monitored during the surgery and recovery periods.

    ➢ Monitoring parameters must be documented at a minimum frequency of every 15 minutes. More frequent documentation of every 5-10 minutes is highly recommended.

    ➢ For more information on medical documentation of surgical procedures and surgical monitoring, please refer to the document “Guidelines for Animal Medical Records for Investigative Personnel” which can be found at http://www.ucuca.umich.edu/guidesurg.htm.

3. Rodent Recovery Surgical Anesthesia and Monitoring

   a) **Pre-surgical:** Once the rodent is anesthetized, the anesthetic depth must be evaluated prior to initiating surgical procedure. Appropriate anesthetic depth is determined by absence of painful response, such as pedal or toe pinch.

   b) **Monitoring:** Monitoring is recommended throughout the anesthetic event every 15 minutes.

    ➢ **Cardiovascular System:**
(i) Tail, foot, tongue or ear color can be monitored for pale or blue 
membranes which is indicative of decreased blood volume, decreased 
perfusion or respiratory distress.

➢ Respiratory System:
(i) Small rodents have respiratory rates too quick to accurately count. 
Instead, an assessment of the breathing pattern should be made. 
(e.g., regular vs. irregular breaths).

➢ Body Temperature:
(i) Rodents have a high body surface area to mass ratio, which causes 
them to heat up quickly when armed and lose body heat quickly when 
chilled. As a result, rodents are prone to overheating when high 
temperature heating systems are used and hypothermia if 
supplemental heat is not provided. Both, thermal excess and thermal 
deficit, can quickly lead to serious injury or death.
• During recovery, heating pads can be placed under the rodent 
cage so half of the cage is on the pad and half off the pad. This 
will allow animals to escape the heat source if they become too 
hot.
• Warming devices should provide gentle heat only (maximum of 
102°F). A thermometer can be placed next to the supplemental 
heat source to verify temperatures are < 102.
• An insulated pad or folded drape must be placed between the 
animal and the heat source.
• AVOID commercial human electric heating pads – maximum 
temperatures are not well regulated and uneven surface 
temperature gradients exist risking thermal injury burns to the 
animal.
• Heating lamps are discouraged since they are a fire hazard risk 
and the maximum temperature is difficult to regulate. However, 
heat lamps can be used as long as they are placed an appropriate 
distance away from the animal to ensure thermal burns do not 
occur.

(ii) Recommended warming devices for rodents
• Circulating water blanket (Gaymar®)
• Rodent isothermal warming pads (Braintree Scientific®)
• Commercial homeo-thermic rodent heating pads with thermostat 
control to regulate maximum temperature
• Commercial insulating pouches and surgical drapes 
(Spacedrapes®)
• Commercial rodent incubators

4. Sedation Without General Anesthesia
a) Monitoring:
➢ Cardiovascular, respiratory, and temperature monitoring guidelines are 
similar to surgical anesthesia monitoring.

b) Oxygen should be supplemented by facemask. For heavy sedation, 
intubation equipment (laryngoscope and endotracheal tube) should be 
available in case protective airway reflexes are lost.

5. Non-Recovery Surgery
a) **Pre-surgical Examination:** A physical examination should be performed including heart rate and rhythm, mucous membrane color (normal = pink), capillary refill time (normal < 2 seconds), respiratory sounds and rate, and temperature. Recommended baseline clinical chemistries will depend upon complexity and length of non-recovery surgery.

b) **Records:** Cardiovascular, respiratory, and body temperature monitoring guidelines are similar to surgical anesthesia monitoring.

6. Additional Monitoring Options: When selecting monitoring equipment, the complexity and associated complications of the surgical procedure (e.g. thoracic, soft tissue, or neurologic) must be considered. In addition, the choice of appropriate monitoring equipment will depend upon the clinical condition and species of the animal. *If surgical procedures will be performed outside of ULAM-ASOR, the ULAM veterinary staff should be contacted for recommendations.*

a) Cardiovascular System:

- **Pulse Oximeter:** Non-invasive method to accurately measure oxygenation (SaPO$_2$) along with pulse rate.
  
  (i) Can be placed on the tongue, ear, skin folds between the pedal digits, nostrils, or commissures of the mouth.
  - Can be placed on the whole paw of small rodents.
  
  (ii) Once placed, confirmation of accurate measurements can be made by comparing heart rate determined by the oximeter with the pulse rate of the animal.
  
  (iii) Placement of the probe on areas of heavily pigmented skin should be avoided as this can cause erroneous results.
  
  (iv) Ideal SaPO$_2$ > 95%
  - SaPO$_2$ < 90% = hypoxia
  - Verify probe placement is correct and investigate signs of hypoxia such as low oxygen tank, hypoventilation, or ventilation-perfusion mismatch.

- **Blood Pressure**
  
  (i) Appropriate cuff size can be accurately determined by gauging the width of the cuff compared to circumference of the limb or tail at the point of application. The cuff width should be 40-50% of the circumference of the limb at that point.
  
  (ii) Doppler ultrasound (Park’s Medical Electronics – Model 811-B, [http://www.parksmed.com/](http://www.parksmed.com/)) in combination with a manual inflatable cuff can be used to obtain systolic blood pressures and will provide continued audible heart rate monitoring.
  
  (iii) Noninvasive Blood Pressure (NIBP) monitors are available for rodents as well as non-rodent mammals. NIBP monitors are typically more expensive than Doppler based probes but provide serial automatic readings without manual manipulations each time.

- **Arterial Blood Gas**
  
  (i) Serial arterial blood gas sampling is the gold standard for evaluating blood oxygenation and ventilation (e.g. PaO$_2$ and PaCO$_2$). Arterial blood gas monitoring is highly recommended for thoracic, cardiovascular, or neurologic procedures.

- **Electrocardiograph (ECG)***
(i) ECGs are recommended if dysrhythmias are expected secondary to surgical procedure or clinical condition. They are useful to assess response to pharmacologic therapy.

b) Respiratory System:
- End-tidal CO$_2$ (ETCO$_2$) – Capnography
  (i) Capnography is useful and recommended for monitoring ventilation of larger species such as pigs, cats, dogs, rabbits, primates and sheep.
  (ii) Ideal range of ETCO$_2$ with anesthesia is 35-50.

c) Procedural Considerations:
- Neuromuscular Blockade – If neuromuscular blocking agents will be administered during a surgical procedure, mechanical ventilation must be utilized. In these instances, heart rate and blood pressure must be monitored and documented every 15 minutes to detect responses to painful stimuli. Animals must be at an adequate depth of anesthesia and non-responsive to surgical stimuli prior to administration of the neuromuscular blockade.
  (i) A peripheral nerve stimulator in conjunction with train-of-four monitoring is highly recommended to monitor the neuromuscular blockade.

E. RELATED DOCUMENTS:
1. Monitoring Forms
   a) ULAM provides the following anesthesia monitoring forms on the UCUCA website: [http://www.ucuca.umich.edu/guidesurg.htm](http://www.ucuca.umich.edu/guidesurg.htm)
      - Non-Rodent Mammal Anesthesia Monitoring Form
      - Rodent Surgical Guidelines – Appendix 1 Rodent Surgical Record
2. Recommended Reading
   a) [AAHA Anesthesia Guidelines for Dogs and Cats](http://www.ucuca.umich.edu/guidesurg.htm)
   b) [AAHA Anesthesia Guidelines for Dogs and Cats](http://www.ucuca.umich.edu/guidesurg.htm)