

# **Mice: Basic Handling and Technique Workshop**

## **University of North Carolina, Chapel Hill**

### **“First do no harm” – Greek Hippocratic Oath, Great Watch Words of Medicine**

#### **Objectives:**

1. Teach methods of safe, humane handling and restraint
2. Teach injection techniques: subcutaneous, intraperitoneal, intravenous and intramuscular
3. Teach blood collection techniques
4. Teach rodent identification methods
5. Teach anesthesia administration and monitoring
6. Teach proper euthanasia methods

#### **Basic Information about working with Mice:**

Proper Personal Protection Equipment (PPE) is a requirement for working with animals. In DLAM facilities, minimum requirements are disposable lab coat, shoe covers, mask and gloves. Please review requirements before entering any animal area! Requirements may change from room to room so each door is posted with instructions. The use of a face mask reduces your risk of allergy to animals. We strongly recommend that you wear masks whenever you work with animals.

#### **If Bitten: *DO NOT PUNISH THE MOUSE FOR ITS NATURAL RESPONSE***

1. Calmly return the animal to its cage
2. Wash the wound with antibacterial soap and water
3. Bandage the wound
4. Notify your supervisor and contact the University Employee Occupational Health Clinic (UEOHC) 966-9119

#### **Mouse Psychology:**

1. Mice are usually mild in temperament and easy to handle. They are not usually aggressive, but can bite if frightened. There are some strains that are aggressive and can inflict painful bites. Mice groom themselves almost constantly to maintain a smooth, glossy haircoat.
2. Mice are nocturnal animals. Activities such as eating, drinking or mating are typically done at night.
3. Dominant mice exhibit a behavior called barbering. Barbering is the dominant mice biting or chewing on the fur of a more subordinate mouse. Barbering should not be confused with fur loss due to illness. Typically, barbering occurs around the face or back.
4. Male mice can be more aggressive and fight more often than females. Aggressive mice should be housed individually to avoid severe injury to cage mates. Generally male littermates may be housed together, but once separated, it is advisable to only house males with females.
5. Mice are creatures of habit. Everyday events do not tend to stress or excite the mice. However, handling and restraint can be stressful and result in the mouse being difficult to work with. Conditioning the mice to such handling (so they do not associate handling and restraint aversively) can make the animals much easier to work with.

## Handling and Restraint:

When picking up adult mice, grasp them gently but firmly at the base or center of their tail. Do not pick them up by the tip of the tail. Place the animal on a surface such as the wire cage top or lid. It is best that the surface not be slick or smooth as mice will behave much more calmly if they have firm footing. While still holding the tail near the base, with your other hand firmly grasp the loose skin on the back starting near the ears using your thumb and first two fingers. The tail can then be held by the last two fingers as shown. Your grip should be firm enough to keep the mouse from struggling, but gentle enough for it to breathe comfortably.



### One-handed Method

Follow the above steps.

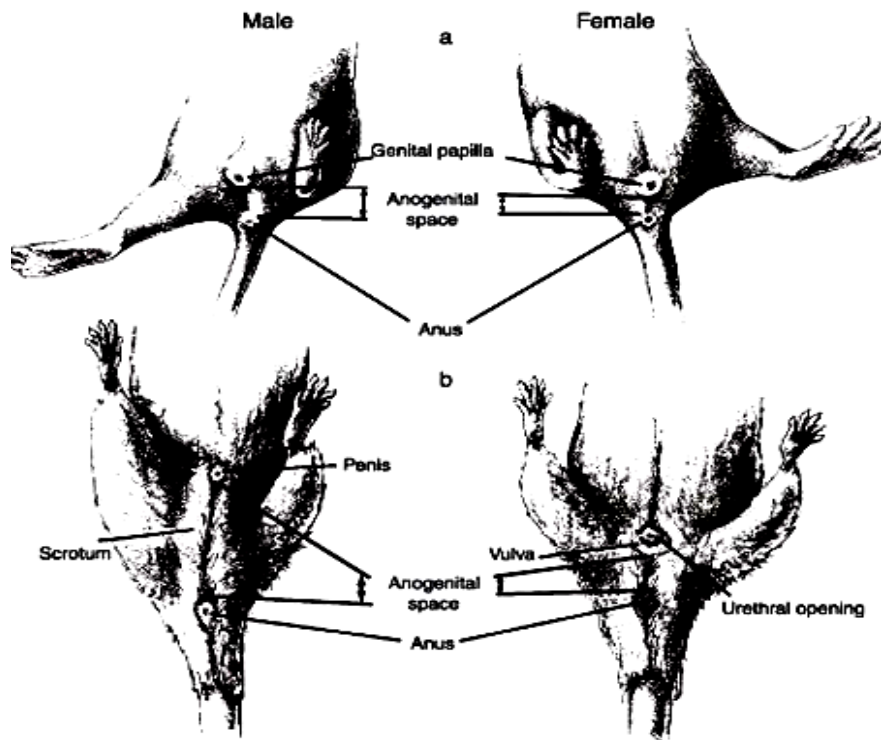
Then, place the mouse's tail between the last two fingers of the hand that is holding the nape.

For quick handling such as cage to cage transfers, it is acceptable to use forceps. Gently grasp the loose skin on the back and quickly transfer them to the new cage. This technique is useful for fractious or aggressive animals. Be sure to clean gloves or forceps with a sterilant such as Spor-Klenz between cages.

Gloves, either light leather, cloth or mesh greatly diminish sensitivity, increasing chance of injury to mice. They also make it more difficult to perform delicate procedures, and mice can often bite through them.

## Sex Determination:

Gender in mice is determined by comparing anogenital distance, or the distance between the urogenital opening and the anus. Male mice typically have a larger anogenital distance when compared with the females. Be aware there are variances in anogenital distance among strains.



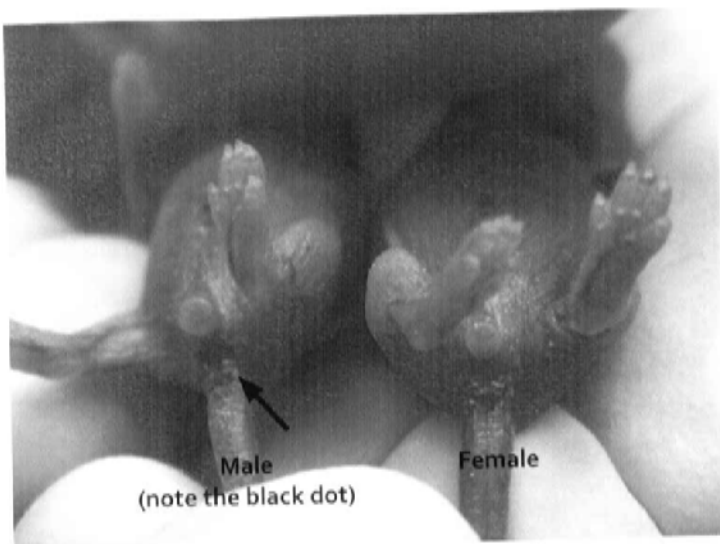
(a) Young Rodent

(b) Adult Rodent

### Tech Tip: Identifying Mouse Genders in Neonates

*By Amanda Ogden, RVT, RLATG, Rodent Breeding Colony Manager, Division of Laboratory Animal Medicine, UCLA*

After several unsuccessful attempts to identify the gender of neonate mice, one of our investigators enlightened us regarding an anatomical feature that could be referenced instead of the routine anogenital distance for the determination of gender. The presence of a black dot at the midpoint of the anogenital divide signifies male gender, and the absence of this dot indicates female. This dot is visible to the naked eye as early as day 0 or 1. Applying this method to roughly 100 neonates, we have had a 100% success rate with black mice, and a 90% success rate with white mice. Sufficient numbers of agouti mouse neonates have not yet been observed to estimate a success rate for this method. This technique has proven to be extremely beneficial for researchers who need to discern gender ratios or who may require specific genders for projects.



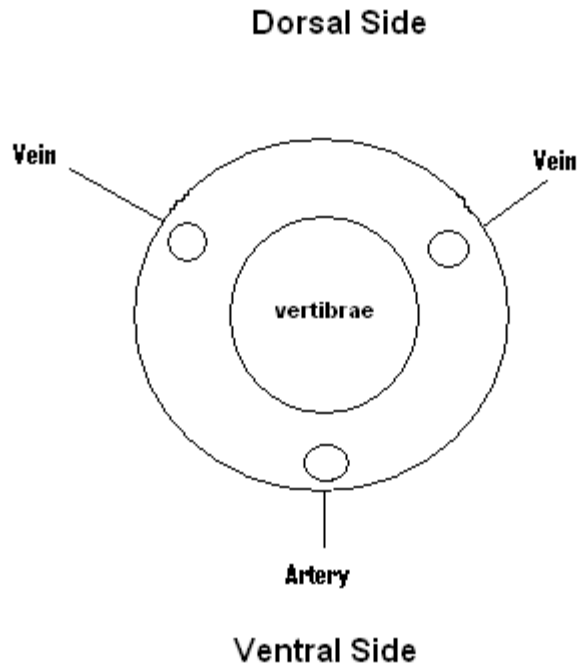
The author wishes to thank Ling Hu and Jacqueline Pleitez for their technical assistance and Jerry Werk for her editorial advice.

## Injections:

With any injection use a fresh, sterile needle for each injection. Do not reuse needles between animals! It is recommended to clean the injection site with a disinfecting agent such as betadine.

1. Intraperitoneal injections – Clean the injection site. Hold the mouse in dorsal recumbency and insert the needle, bevel up, in a position midway between the bend of the knee and the midline. Avoid the midline region to ensure that the bladder is not punctured. The needle should be held at an approximate 45 degree angle with the body. Ideal needle size is 23 gauge or smaller. [\*Amounts of 2-3 mL can be administered, depending on the size of the mouse\*](#)
2. Subcutaneous injections – Clean the injection site. The injection may be performed in any area of loose skin along the back or flank. One method is to tent the skin between the shoulder blades and insert the needle bevel up in the pocket created. Aspirate to be sure you have not entered a blood vessel. If there is no blood in the hub of the needle, inject. A second method is to insert the needle just under the skin along the mouse's flank. Pull the needle up flat against the skin. If the outline of the needle is clearly visible it is subcutaneous and not in the body cavity. Aspirate and inject. Ideal needle size is 23 gauge or smaller. [\*Amounts of 2-3 ml can be injected\*](#) into a large mouse; however, the injection should not make the skin at the site taunt. It is best to inject a large volume in several sites along the back
3. Intramuscular injections – Clean the injection site. The muscle mass running along the back of the leg is best used. Insert the needle bevel up at an angle parallel to the femur (avoiding the sciatic nerve). Aspirate; if there is no blood in the needle hub inject slowly. The ideal needle size is 27gauge. Note that this is a potentially painful injection. If any other injection such as subcutaneous or intraperitoneal can be used, avoid intramuscular injections. [\*MAX: 0.05-0.1mL may be injected per injection site.\*](#)
4. Intravenous injections – Dilate the blood vessels by warming the mouse. Placing the mouse under a heat lamp for a short period (2-5 minutes) works well. Watch the mouse closely to avoid overheating. Once warmed, place the mouse in a plexiglass restrainer. Clean the tail. Locate one of the lateral veins. Insert the needle bevel up toward the base of the tail. Note that the vein is very superficial and easily passed through. If the needle is properly seated, inject slowly, the vein will clear from the injection to the base. If the needle is not properly seated you will note ballooning at the injection site. Stop immediately and try again further toward the base of the tail. The ideal needle size is 27 gauge or smaller. [\*Amounts of 0.2ml may be given in this manner.\*](#)

## Cross Section of Mouse Tail



### Oral Gavage:

We recommend using stainless steel, ball tipped gavage needles. It is important to pre-measure the needle before gavaging. The tube should measure the distance from the tip of the nose to the last rib, so that the needle will pass down the esophagus into the stomach. If the tube is too short, the injected fluid may be aspirated by the mouse causing possible pneumonia and death.

The mouse should be firmly restrained. Insert the gavage needle into the mouth at one side allowing the mouse to chew on it. Slide the needle down the back of the throat while tilting the mouse's head back, so that the neck is in a straight line. The needle should pass easily down the esophagus; with little to no resistance. If the mouse struggles or resistance is met, stop, back up and start over. Improper gavage technique can cause tearing of the esophagus or asphyxiation.

Observe the mouse carefully after the gavage is completed. No fluid should be coming from the mouth or nose and the mouse should not show signs of distress.

The following table indicates appropriate needle sizes:

<b>Mouse Wt (g)</b>	<b>Gauge</b>	<b>Length (inches)</b>	<b>Ball Diameter (mm)</b>
Up to 14g	24	1	1.25
15-20g	22	1 or 1.5	1.25
20-25g	20	1, 1.5 or 3	2.25
25-30g	18	1, 1.5 or 2	2.25
30-35g	18	2 or 3	2.25

### **Various Helpful Tips:**

1. When collecting blood from laboratory animals, the largest recommended amount is 1.5% of the animal's total body weight. Collection should not occur again for two weeks. This will allow blood constituents to return to normal. If blood is needed weekly, 0.5% body weight is a safe amount.
2. When giving substances intravenously, inject slowly to avoid shock.
3. Injecting slowly when giving substances intramuscularly will cause the least amount of pain.
4. The volume of blood in a mouse is about 78-80 ml/kg. This is approximately 10% of its body weight. Only about half of this can be recovered in a terminal blood withdrawal procedure.
5. Remove surgical clips or nonabsorbable suture within seven to ten days after surgery to avoid skin overgrowth.
6. Check rodents' teeth frequently. This will insure early detection of malocclusion. If maloccluded, teeth may become overgrown and interfere with eating.

### **Acknowledgements:**

The University of North Carolina would like to thank:

The AALAS Learning Library  
University of Minnesota, Research Animal Resources  
University of Texas Medical Branch at Galveston

### **Training Information:**

For additional training please contact the Training and Compliance Coordinators for the Institutional Animal Care and Use Committee (IACUC) at 966-5569. We offer training in both one on one and classroom settings.

For a look at IACUC Guidelines and dates of future classes, visit our website at [www.research.unc.edu/iacuc/](http://www.research.unc.edu/iacuc/).

## RODENT IDENTIFICATION

There are several IACUC approved methods used to identify rodents. Each method has both advantages and disadvantages. In long-term studies, it is important to choose a method that is permanent and easily read.

### Long Term Methods:

- **Ear Notching** – This method is frequently used in both mice and rats. There are several tools that may be purchased to achieve this. Most resemble a hole puncher and are very cheap. There are previously created maps that serve as a numbering system, or the researcher may create a map.
  - A) **Advantages** – Ear notching can be done quickly while causing very little pain or distress. The instruments are not costly and can be obtained easily.
  - B) **Disadvantages** – This method can not be applied until the ears are fully developed. This may be too late for those that use young rodents. This may not work with fractious strains. Rips or tears caused by fighting may leave the pattern indiscernible. Tools used to notch ears dull easily so must be replaced frequently.
  
- **Ear Tagging** – Ear tags can be purchased with numbers and/or letters. Correct placement of the tag makes them fairly easy to read.
  - A) **Advantages** – Ear tags are inexpensive and are fairly easy to apply. This method does not require the use of anesthesia. Tagging can be done quickly and does not seem to cause pain and only minor distress.
  - B) **Disadvantages** – Tags can fall out if not applied properly. They can also be lost if ears are ripped or torn in strains that fight. Different sized tags are available for different species. Tags are relatively heavy for weanlings and may cause young mice to tilt their head even when the proper sized tag is applied. Some strains are prone to scratching the tagged area which can lead to infection, hematomas, and granulomas. In a very limited number of cases, a member of the DLAM veterinary team has seen ear tags stimulate tumor growth.
  
- **Microchipping** – Microchips, electronic transponders, are safe and reliable.
  - A) **Advantages** – Microchips may be applied without the use of anesthesia. Applying microchips seems to cause little or no pain. Even though the chip may migrate to a different area, they are not lost so prove to be a reliable method. Animals can be identified without handling and removing them from the cage. Some microchips are designed to provide other information such as core body temperature and heart rate.
  - B) **Disadvantages** – The equipment used to read the chips is fairly expensive. Microchips cost five to ten dollars each. Despite the manufacturer's recommendation, chips can be reused. In order to reuse the chips they must be sterilized by ethylene oxide. The hospital will do this as a service for fee. If not implanted properly there is a slight risk of infection. In a very limited number of cases, a member of the DLAM veterinary team has seen microchips stimulate tumor growth.

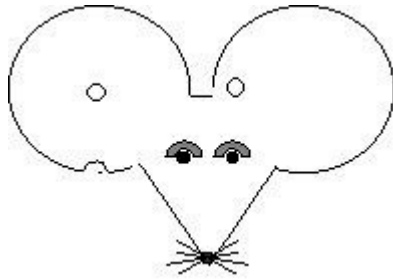
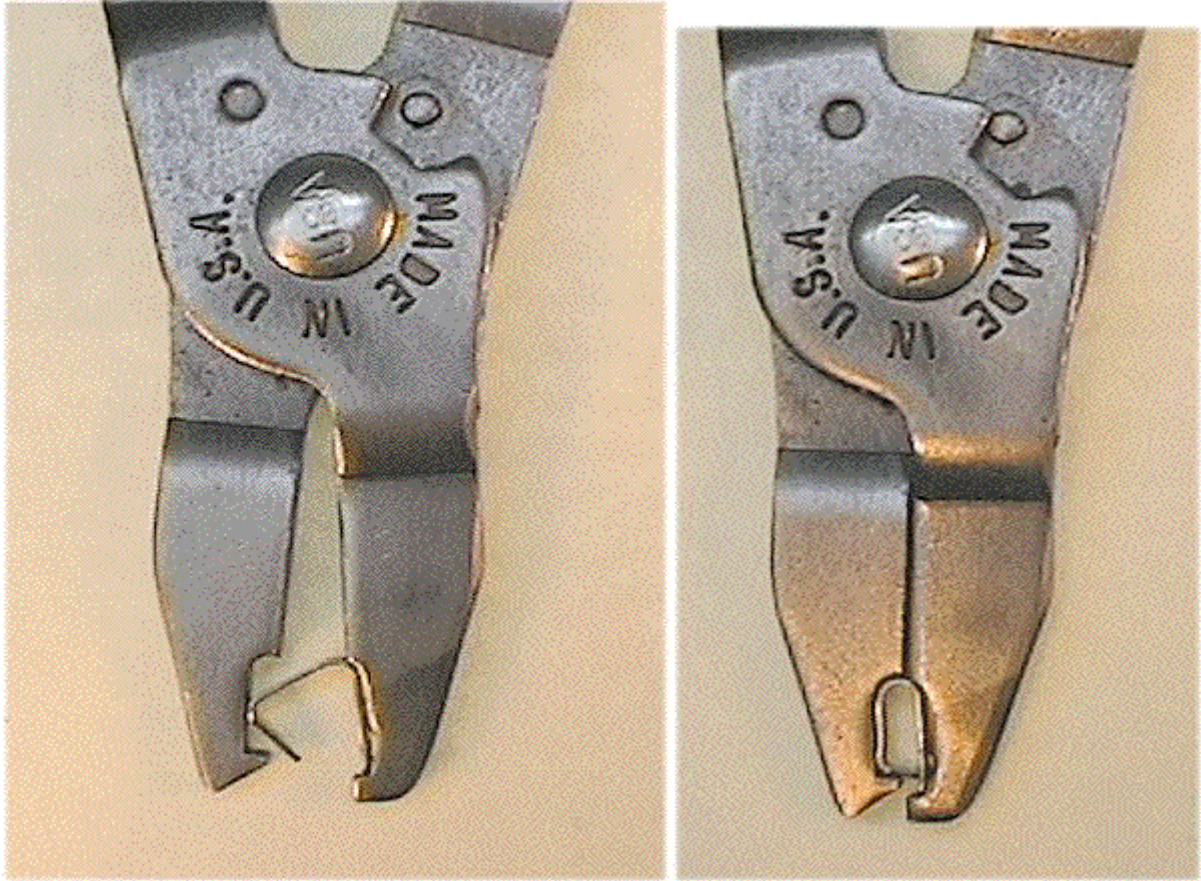
- **Micro tattooing** – This method seems to be growing in popularity. It is both permanent and fairly easy to apply.
  - A) Advantages** – This method works in all strains, even the more fractious. The cost is very reasonable after the initial expense. Tattoos can be applied to rodents of any age. The markings are easily read, especially when applied to the tail of light colored rodents. When placed in the proper area, it is not necessary to handle the animal to read the tattoo. Tattooing causes only minor pain and distress and does not require the use of anesthesia.
  - B) Disadvantages** – The identifying marks may be a little difficult to read in young pigmented mice. This improves as the mice age. The initial cost is rather expensive. There is a small chance of inducing infection if the tattoo is not applied correctly.
  
- **Toe Clipping** – “Toe-clipping, as a method of identification of small rodents, should be used only when no other individual identification method is feasible and should be performed only on altricial neonates.”<sup>1</sup> The IACUC allows a maximum of four toes and no more than two per foot. Do not cut the hallux (“dew-claw” or “little toe”) as this may decrease the rodent’s grasping ability.
  - A) Advantages** – Toe clipping can be done at a very early age, post natal day one. The ideal time is between post natal day five and seven when the toe is large enough to work with yet the bones are not calcified. The tissue can be used for genotyping. Toe clipping may not be performed after post natal day ten. No anesthesia is needed. This seems to cause little or no pain when performed early enough. The young react to being removed from their mother, but do not react to the clipping of the toe.
  - B) Disadvantages** – The young show signs of distress when removed from their mother and siblings. This may cause a small amount of pain. The general public views this as a form of brutal mutilation. There is a small possibility of infection. A reduction in the number of toes may reduce the ability to grasp objects.

## **Short Term Methods:**

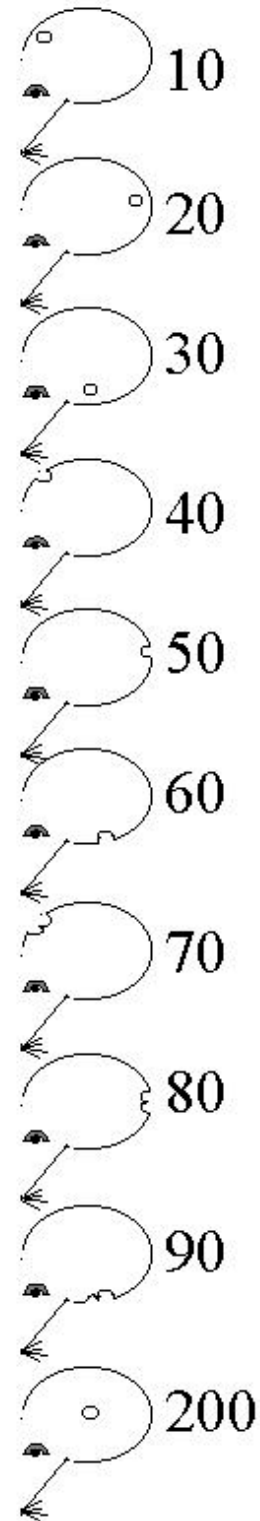
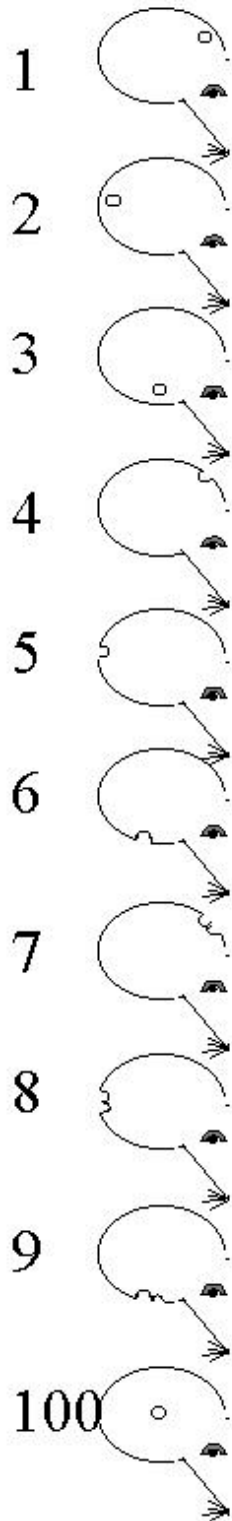
- **Hair Clipping** – Trim patterns into the fur. Keep a record or picture to identify rodents.
  - A) Advantages** – Causes no pain.
  - B) Disadvantages** – This is very temporary. The hair will grow back within ten days and the clipping must be repeated.
  
- **Permanent Markers and Fur Dyes** – It is easy to apply marks or dyes to different body parts.
  - A) Advantages** – This method is non-invasive. It causes only minor stress due to restraint.
  - B) Disadvantages** – This method can be time consuming, since it must be repeated soon after the previous marking. If working with nursing pups, the mothers will groom the neonates excessively and the markings may disappear overnight. This could result in a loss of identity.

**If you would like to inquire about equipment used in methods discussed above, please email the OACU Training/Compliance Team. You may send questions to the general IACUC email account [iacuc@med.unc.edu](mailto:iacuc@med.unc.edu)**

<sup>1</sup>*Guide for the Care and Use of Laboratory Animal Care and Use.* National Research Council. 1996. National Academy of Sciences. 46 pp.



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## **ACCEPTABLE METHODS OF RODENT BLOOD WITHDRAWAL**

PDF format available at:

<http://research.unc.edu/iacuc/sop/documents/BloodWithdrawal-Rodents.pdf>

## **INSTITUTIONAL ANIMAL CARE AND USE COMMITTEE** **RAT AND MOUSE EUTHANASIA POLICY**

Performing euthanasia correctly is an ethical imperative. Proper euthanasia is quick, minimizes pain/distress and reliably causes death. Practical issues such as degree of technical difficulty, time required to perform the procedure and readily available equipment/resources to perform the procedure as well as aesthetics and human emotion must also be considered. Standardized guidelines for humane euthanasia have been detailed in the June 2007 AVMA Guidelines on Euthanasia: [http://www.avma.org/issues/animal\\_welfare/euthanasia.pdf](http://www.avma.org/issues/animal_welfare/euthanasia.pdf)

### **Euthanasia of animals**

Animals may be euthanized by trained research staff, or they can be labeled for euthanasia which will be performed, with a fee for service, by Division of Laboratory Animal Medicine (DLAM) staff. [However see below for sick or injured animals]. When requesting DLAM service, research personnel must do the following: 1) complete and submit a "Request for Euthanasia in Animals" form (available at <http://research.unc.edu/dlam/downloadhusforms.htm>) and leave the animal(s) in the cage, and 2) place a euthanasia card on the cage so that DLAM can readily identify the animal(s) slated for euthanasia. Animals should always have food and water. Unless there is proper documentation on a euthanasia request form, the investigator is responsible for any errors or miscommunications that may occur during the euthanasia process. Do not make verbal arrangements with DLAM staff.

Young animals that are still nursing must not be removed from the lactating female. Leave the dam with the young, place a euthanasia card on the cage, and fill out a request for euthanasia form stating that only the pre-weanling animals are to be euthanized. Alternatively properly euthanize the young yourself.

All animals slated for euthanasia must be housed according to housing standards of UNC-Chapel Hill and the *Guide for the Care and Use of Laboratory Animals*. Cages marked for euthanasia must not be overcrowded. Do not stack cages slated for euthanasia on top of each other. This blocks air flow into the cage. **All personnel performing euthanasia are responsible for making sure that euthanasia is confirmed.**

### **Humane euthanasia of sick or injured animals**

Sick or injured animals may require humane euthanasia if successful treatments are unlikely or if pain and distress cannot be appropriately managed. Moribund animals must also be euthanized in a timely manner. When sick, injured or moribund animals require euthanasia, it must be performed immediately; animals cannot be held for later euthanasia by DLAM personnel.

DLAM veterinary staff has authority to euthanize moribund animals or animals experiencing more than momentary or slight pain or distress. If DLAM veterinary staff is unable to contact research personnel regarding the care or treatment of a moribund animal, DLAM is authorized to euthanize the animal.

## MOUSE AND RAT EUTHANASIA

### Section 1

#### Terms and Definitions

The following terms and definitions detail the UNC-Chapel Hill methods for conducting humane euthanasia in rats and mice. Exceptions must be described in an animal care and use protocol and reviewed and approved by the Institutional Animal Care and Use Committee (IACUC) on a case by case basis.

#### Terms and Definitions

**Physical methods of euthanasia**—to ensure animals are dead, one of the following procedures must be performed on animals that have been anesthetized with approved agents: 1) cervical dislocation; 2) decapitation; 3) thoracotomy (open the chest cavity using sharp scissors or scalpel); or 4) collection of vital organs may be performed if tissues are needed for experimental purposes. Note: these physical methods can only be performed by personnel who have been properly trained. Cervical dislocation cannot be performed on rodents that weigh greater than 200 grams.

Physical euthanasia performed on unanesthetized animals requires that all individuals performing the procedure are observed and certified by approved IACUC or DLAM staff members. Please note that Laboratory Coordinators **MAY NOT** certify persons performing physical euthanasia on unanesthetized animals. Performing physical euthanasia on unanesthetized animals can only be done if the procedures are described in an approved animal care application (ACAP).

**Decapitation**—decapitation in unanesthetized neonatal and adult rodents is permitted only if it is performed correctly by a trained person, its use is scientifically justified, and it is approved by the IACUC in the animal use application. When performed properly this technique is nearly instantaneous and is considered humane. Guillotines that are designed to accomplish decapitation in adult rodents in a uniformly instantaneous manner are commercially available. Sharp scissors can be used to decapitate neonatal rodents. Check guillotine and scissor blades frequently to ensure sharpness. “The equipment used to perform decapitation should be maintained in good working order and serviced on a regular basis to ensure sharpness of blades. The use of plastic cones to restrain animals appears to minimize stress from handling, minimize the chance of injury to personnel, and improves positioning of the animal in the guillotine.” ([2007 AVMA Guidelines on Euthanasia](#)) Note: The Physics Department’s Instrument Shop, located in Phillips Hall 115A, will sharpen blades for a small fee.

**Cervical Dislocation**—cervical dislocation in unanesthetized neonatal and adult rodents is permitted only if it is performed correctly by a trained person, its use is scientifically justified, and it is approved by the IACUC in the animal use application. Manual cervical dislocation is a humane method of euthanasia when limited to rodents weighing less than 200 g and when performed properly by a certified person with a high degree of technical competency. Personnel using cervical dislocation must be adequately trained, demonstrate their technical proficiency, and must consistently apply this method humanely and effectively.

**Gaseous carbon dioxide (CO<sub>2</sub>)**—used in rodent euthanasia: CO<sub>2</sub> must be supplied using a compressed gas tank. The use of dry ice as a source of CO<sub>2</sub> for euthanasia is not permitted.

**Inhalant anesthesia**—anesthetic agents that are delivered as a volatile gas to the respiratory tract and when used properly, effectively induce anesthesia. Personnel should minimize their exposure to these agents as several can be harmful (potentially causing liver damage or birth defects if pregnant women are exposed). These agents should only be used in a fume hood, ducted biosafety cabinet or in a system with an active gas scavenging device.

**Injectable anesthesia**—chemicals that induce anesthesia when injected with needle and syringe: common routes of injection include intraperitoneal (IP), intramuscular (IM) or intravenous (IV). Advantages include the following: easy to perform, minimal equipment, and avoids safety concerns associated with inhalants.

## Section 2

### Procedures

#### Gaseous Carbon Dioxide

<http://research.unc.edu/iacuc/sop/documents/CarbonDioxidePosting.pdf>

#### Inhalant Anesthetics (e.g. Isoflurane)

Induction chambers for inhalational anesthetics must be of appropriate size so as not to overcrowd animals but not so large as to require large volumes of the anesthetic agent which slows induction time. Induction chambers must have a perforated platform to place the animals on to prevent direct contact with anesthetic soaked gauze (or other absorbent material). The lid should be tight fitting and the chamber must be used in a fume hood, a ducted biosafety cabinet, or with a properly functioning active scavenging system.

1. Pre-charge the anesthetic chamber by placing two to three pieces of gauze on the bottom of the chamber, below the perforated platform. Add approximately 5 ml of Isoflurane liquid to the gauze. Close the lid and wait 5 minutes for the liquid to form a volatile gas within the chamber. (Note: the amount of Isoflurane to add depends on the size of the chamber, so the actual amount needed may vary.)
2. Remove the lid of the chamber and quickly place the animals onto the perforated platform within the chamber and immediately close the lid.
3. The animals should become anesthetized in 2-5 minutes. Neonates must remain in the chamber for at least five (5) minutes.
4. When animals are completely recumbent and are obviously deeply anesthetized remove them from the chamber.
5. **Immediately** perform a physical method of euthanasia. Isoflurane is highly volatile and animals will quickly regain consciousness once removed from the chamber; thus it is imperative that physical euthanasia be performed immediately.

## Injectable Anesthetics

Injectable anesthetics can be effectively used to anesthetize animals prior to performing physical euthanasia. The agent should be a standard drug for the species and the dosage used should be equal to or greater than standard published reference dose for anesthesia (e.g., a common dose of pentobarbital for euthanasia is 100 mg/kg, which is approximately twice the anesthetic dose for rats and mice). Once animals are properly injected (usually IP), allow sufficient time for them to lose consciousness.

Injectable anesthetics intended for use in adult rodents may not have the desired effect in neonates. In a pilot study conducted at UNC-Chapel Hill, few anesthetics were found to be reliably effective in neonates. The drugs that provided the most effective anesthesia are available only to veterinarians and as a result were considered impractical for use by the scientific community. Contact a DLAM veterinarian for more information about appropriate doses of injectable anesthetics.

## Fetuses

Fetuses **up to 14 days** in gestation: neural development at this stage is minimal and pain perception is considered unlikely. Euthanasia of the mother or removal of the fetus should ensure rapid death of the fetus due to loss of blood supply and non-viability of fetuses at this stage of development.

Fetuses **15 days** in gestation to birth: the literature on the development of pain pathways suggests the possibility of pain perception at this time. Whereas fetuses at this age are not sensitive to inhalant anesthetics, euthanasia may be induced by the skillful injection of chemical anesthetics. Decapitation with surgical scissors and cervical dislocation are acceptable physical methods of euthanasia when used by a trained person. When chemical fixation of the whole fetus is required, fetuses should be anesthetized prior to immersion in or perfusion with fixative solutions. Anesthesia may be induced by hypothermia of the fetus, by injection of the fetus with a chemical anesthetic, or by deep anesthesia of the mother with a chemical agent that crosses the placenta, e.g., pentobarbital. Consult with one of the institutional veterinarians to learn more about fetal sensitivity to specific anesthetic agents. When fetuses are not required for study, the method chosen for euthanasia of a pregnant mother must ensure rapid death of the fetus.

## Euthanasia References

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American Veterinary Medical Association (2007) AVMA Guidelines on Euthanasia. [http://www.avma.org/issues/animal\\_welfare/euthanasia.pdf](http://www.avma.org/issues/animal_welfare/euthanasia.pdf).

Institute of Laboratory Animal Resources Commission on Life Sciences, National Research Council (1996) Guide for the Care and Use of Laboratory Animals. National Academy Press (65-66).

NIH (2002) Guidelines for the Euthanasia of Rodent Feti and Neonates.

## Supply and Vendor Information

- Braintree Scientific                      781-843-2202                      [www.braintreesci.com](http://www.braintreesci.com)  
Instruments, lab equipment, isothermal pads, tattoo paste
- Fisher Scientific                          800-766-7000                      [www.fishersci.com](http://www.fishersci.com)  
Lab equipment, chemicals, instruments, pharmaceuticals
- Henry Schein                              800-872-4346                      [www.henryschein.com](http://www.henryschein.com)  
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Need Vet License
- Kent Scientific                            888-572-8887                      [www.kentscientific.com](http://www.kentscientific.com)  
Surgical equipment, telemetry equipment
- Med-Vet International                    800-544-7521                      [www.shopmedvet.com](http://www.shopmedvet.com)  
Veterinary supplies and instruments (discounted)  
Need Vet License
- National Band and Tag                  859-261-2035                      [www.nationalband.com](http://www.nationalband.com)  
ID tags, ear tags
- Roboz                                        800-424-2984                      [www.roboz.com](http://www.roboz.com)  
Specialize in instruments
- TW Medical                                888-787-4487                      [www.twmedical.com](http://www.twmedical.com)  
Veterinary supply (Bill Forrester)
- UNC-CH Materials Management and Distribution      966-5671  
Scientific Storeroom, General Storeroom, Chemical Storeroom
- Veterinary Medical Supply 800-533-8674  
Veterinary Supplies out of Zebulon, NC  
Need Vet License
- Southern Anesthesia                    800-456-0757                      <http://www.sasvet.com/>  
This is a human source company that has a Veterinary division, will set up an account  
without a vet license.

# The University of North Carolina at Chapel Hill

## IACUC Training Record

### Mice: Handling and Basic Techniques

Performed?	Technique	Comments
	Handling and Restraint	
	Intraperitoneal Injection	
	Subcutaneous Injection	
	Ear Notch	
	Ear Tag	
	Intramuscular Injection	
	Intravenous Injection	
	Tail Nick Bleed	
	Tail Clip Bleed	
	Oral Gavage	
	Submandibular Bleed	
	Anesthesia, administration	
	Anesthesia, monitoring	
	CO <sub>2</sub> w/ Phys. Euthanasia	
	Cardiac Puncture, terminal	
	Retro-orbital Bleed	*Individual appointment during class*
	Cervical Dislocation No Anesthesia	*Individual appointment during class*
	Sexing	*OACUC will teach this technique once your colony is ready*
	Saphenous Bleed	*By individual appointment only*
	Isoflurane Vaporizer Training	Contact DLAM Vet Services at 966-2906
	Other:	

**Please add email address if you are a Lab Coordinator:** \_\_\_\_\_

**Phone #:** \_\_\_\_\_

**I certify that I have received the above training:**

Signature: \_\_\_\_\_

Print Name: \_\_\_\_\_

Instructor Signature: \_\_\_\_\_

PID: \_\_\_\_\_

PI: \_\_\_\_\_

Date: \_\_\_\_\_