LAB_093 Urinary Catheter Insertion in Female Mice (Expiry: May 2029)

I. OBJECTIVE

To perform urinary catheter insertion in female rodents to deliver substances into the bladder.

NB: The use of (*) indicates this statement is dependent on the facility procedures
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II. DEFINITIONS

Competent – "the consistent application of knowledge and skill to the standard of performance required regarding the care and use of animals. It embodies the ability to transfer and apply knowledge and skill to new situations and environments." [1]

III. COMMENTS / RECOMMENDATIONS

- Users must keep monitoring records, which includes surgical records (example templates can be obtained by contacting the UQBR Veterinarians or Animal Ethics Unit Veterinary Officer).
- Any associated experimental compounds or medications (including your anaesthetic protocol) must be detailed within the Animal Ethics Committee (AEC) application.
- PPE is facility dependent; however, this should at least include disposable gloves, long sleeved lab gown, face mask, safety glasses, hair bonnet, closed in shoes.
- Active heating (e.g. a heat mat) must be used. *
- Clean surgical technique must be, practised as per LAB_002 *Clean Technique for Laboratory Animal Surgery*
- Wherever practicable, aseptic surgical technique must be practised as per LAB_001 *Aseptic Technique for Laboratory Animal Surgery*
- Adverse events must be managed as per LAB_022 *UQBR Managing Adverse Health Events* and reportable events submitted to the AEC within 7 days.

IV. EQUIPMENT

- Disinfectants: surface disinfectant (e.g. 70% ethanol) and skin disinfectants (e.g. chlorhexidine based). Refer to LAB_001 *Aseptic Technique for Laboratory Animal Surgery* and LAB_002 *Clean Technique for Laboratory Animal Surgery* for options. *
- Clean recovery boxes – standard housing boxes including sterile feed, water, appropriate nesting materials (to aid thermal support) and environmental enrichment.
- Active heating equipment (e.g. fit for purpose heat mats, Aria Ventilated Cabinets®) * as per LAB_058 Heating Procedures in Mice and Rats
- Anaesthetic agents and equipment**
- Ophthalmic lubricant (non-medicated, viscous and pH neutral: e.g. Refresh "Lacri-lube"©, Visco-tears© gel)
- Sterile surgical consumable
    - Including: sterile gauze, sterile cotton tips, IV catheter (24G x ¾") (0.47 x 19mm)

V. PROCEDURE

1. Prepare yourself and the workstation as per LAB_001 *Aseptic Technique for Laboratory Animal Surgery* / LAB_002 *Clean Technique for Laboratory Animal Surgery*
2. Prepare clean, warm recovery boxes (e.g. resting on a heat mat).
3. Anaesthetise the animal**
4. Check for the absence of a withdrawal reflex. If a withdrawal reflex is present, the animal is not sufficiently anaesthetised and anaesthetic depth needs to be increased prior to proceeding.
   If movement of skeletal muscle, or withdrawal reflexes are present at any point throughout the procedure, activity must stop and only resume once sufficient anaesthetic depth regained. If you are having difficulty maintaining appropriate anaesthetic depth consult a UQBR veterinarian (once the animal has recovered, and before proceeding to anesthetise any more animals).
5. Apply ophthalmic lubricant to both eyes, using a sterile cotton tip.
6. Lie the rodent in dorsal recumbency and express the bladder by manual palpation using gentle pressure.
7. Prepare the animal by disinfecting the peri-urethral area as per LAB_001 *Aseptic Technique for Laboratory Animal Surgery* / LAB_002 *Clean Technique for Laboratory Animal Surgery* but ensuring that alcohol is not used on mucous membranes like the urethra.
8. Gently insert the catheter into the urethra until it reaches the bladder using one of the below methods.
   Correct placement is indicated by smooth advancement of the catheter with minimal resistance, and the immediate or gentle flow of urine from the catheter once it reaches the bladder.
   The catheter is hydrophilic so along with the lubrication from the Betadine/PBS used for disinfection should slide easily into the urethra. The urethra has a 'U' bend from the external opening to the bladder. Understanding the anatomy will make catheter insertion much easier. Some rodents may be difficult to catheterise, if this is the case identify the animal on the cage card to avoid its future use (for this procedure). Ensure a new sterile catheter is used for each animal. As a guide, the correct length of insertion for a 7-week-old female is 10mm, a warning mark on the catheter is useful.
   For training purposes, the use of fresh white mice cadavers and pigmented water-soluble solution is helpful as the catheter and correct placement in the bladder can be visualized. A volume of 25ul is sufficient to measure successful delivery.

Method One

a. Restrain the hips firmly and push skin/peri-urethral area caudally (towards the tail) to straighten the urethra.
b. Insert tip of catheter, then slightly reduce hold on the peri-urethral area.
c. Gently advance the catheter to the bladder, reducing hold on peri-urethral to improve movement of catheter under the pubic bone.

Method Two

a) Insert tip of catheter in a cranial to caudal (head to tail) direction about 3mm.
b) With the tip in place swing the free end of the catheter caudally to the direction of the tail tip until it is parallel with the tail.
c) Gently advance catheter to the bladder.

Method Three

a. Place the animal in ventral recumbency (on its front). Lift the animal by the tail and bend the tail backwards until perpendicular to spine and flexing the spine over your hand.
b. Insert the catheter tip and then maintain a slight curve in the catheter as it is advanced to the bladder.

Figure 1. Urinary catheter

Figure 2 Placement of a urinary catheter in a female mouse, demonstrated in a cadaver. .

9. Once the catheter is in the correct location push slowly on the plunger to deliver the solution. Up to 50uL may be injected.
If resistance is felt withdraw slightly to re-position as the catheter tip may be pushing against the wall of the bladder.

10. Slowly remove the catheter at a 30–45-degree angle to reduce leakage.

11. Place the animal into a recovery box, maintained on a heat mat until fully ambulatory. If available, recovery boxes may then be placed into a climate controlled, Ventilated Cabinets® for ~12 hours recovery.

12. Continuously monitor (keep within eyesight) all mice during surgery and throughout the recovery phase until fully ambulatory.
It is common to observe grooming of the per-urethral area up to 30 minutes following recovery.

13. Clean and disinfect all equipment between each animal.

VI. BIBLIOGRAPHY

1. NHMRC. (2013). Australian code for the care and use of animals for scientific purposes, 8th edition. National Health and Medical Research Council (NHMRC). Retrieved from https://www.nhmrc.gov.au/about-us/publications/australian-code-care-and-use-animals-scientific-purposes.

2. St Clair, M, Sowers, A, Davis, J, & Rhodes, L 1999, 'Urinary Bladder Catheterization of Female Mice and Rats', *Contemporary Topics in Laboratory Animal Science,* vol. 38, no. 8, pp. 98-79.

Version #	Reviewing AEC	AEC Review Date	Outcome
2.0	Anatomical Biosciences AEC, Health Sciences AEC, Molecular Biosciences AEC and Laboratory Biomedicine AEC	16/02/2022 12-month extension approved March 2025	Approved to March 2026
3.2	Laboratory Biomedicine AEC	07/05/2026	Approved to 07/05/2029