Scisense Pressure Surgical Protocol

Rat Intracranial Pressure (ICP) Measurement

APPLICATION BASICS

Site:	Intracranial space (3 mm into parenchyma)
Species:	Rat
Body Weight:	400 - 650 grams
Duration:	Acute

CATHETER

Size:	1.6F
Type:	Pressure
Catalog #:	FTH-1611B-0018
SYSTEM	SP200, SP430, ADV500

Application

The measurement of Intracranial pressure (ICP) is an invasive procedure to determine the pressure inside the cranium. The cranium limits expansion capabilities of the brain; therefore, as volume increases in the cranium the ICP will also increase. Precise monitoring of pressure within the cranium can be valuable for a variety of applications, such as:

- Stroke
- Tumors
- Subarachnoid hemorrhage
- Severe brain injury
- Coma of unknown etiology
- Reye syndrome
- Hydrocephalus

Studies suggest improved clinical assessment from ICP waveform analysis. Specifically, ICP pulse pressure, measured by an invasive solid-state pressure micro-manometer, offers improved outcome when ICP waveforms are used as a treatment management target.

Pre-Surgical Preparations and General Anesthesia

See Research Equipment Sources (RL-90-tn) for recommended equipment suppliers. Prepare an area for scrubbing in a separate location from where the surgical operation will take place. For intracranial surgery, it is best to find low-traffic area. Ideally, clean surfaces using disinfectants with low reaction to organic materials (e.g. Phenolics - Lysol, TBQ).

Basic surgical supplies for rat intracranial surgery should include a sterile surgical instrument pack and sterile supplies (i.e. drapes, 4 x 4" gauze squares, Q-tips, disposable high-temp fine tip cautery, 5 ml syringes, saline rinse, tray, gloves, mask, head bonnet and sterile suture packs). In addition, a glass bead sterilizer, heating water blanket or approved electrical heating/feedback control unit should be used. Heat lamps are not ideal for body temperature maintenance and can often be a source of electrical noise/interference. Delicate rodent surgical instruments should be inspected for damage before sterilizing.

Set up surgical microscope (interpupillary distance, check light bulbs, adjust to check magnifications), organize surgical table and fine-tune surgical stool to a comfortable setting where the triangular position can be reached (both feet touching the ground with both arms comfortably resting on the surgical table). Turn on glass bead sterilizer.

Prepare 0.9% saline or a similar isotonic fluid and pre-warm the solution if it will be given pre-operatively. When a decision is made to use pre-warmed sterile isotonic fluids subcutaneously it is also suggested to use a preventive analgesia.

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Rat Intracranial Pressure (ICP) Measurement Cont.

Pre-Surgical Preparations and General Anesthesia Cont.

Before inducing anesthesia be sure to record weight, age, sex, strain, colony history and health status of each rat, and determine whether animals have had enough acclimatization time (usually 3 days post arrival).

Please adhere to your institutions guidelines for anesthesia and pain management. For reference on the methods of anesthesia or analgesia please refer to Rodent Anesthesia Guidelines (RL-67-tn). Following the induction of anaesthesia, shave the animal while on the warming pad using a #40 blade attached to Oster Small Animal Clippers (Harvard Apparatus). Furthermore, remove any remaining hair from the surgical area using a depilatory cream (e.g. Nair). For recovery procedures, apply surgical scrub alternating between disinfectant (i.e. iodophores, chlorhexidines) and alcohol. Please remember: Iodophores will inactivate a wide range of microbes, however literature describes their reduced activity in the presence of organic matter.

Use gauze squares for scrubbing. Scrubbing should always begin along the incision line and extend outwards, ensuring contaminants are not pulled towards the surgical site. Always scrub a larger surface area than the surgical field. Do not wet a large area of skin or fur with alcohol as this can cause hypothermia. Finally, consider using drapes to maintain a sterile field and preserve body temperature.

Surgical Approach

FEMORAL ARTERY CATHETERIZATION

Once stable surgical anaesthesia is confirmed, secure animal in dorsal position (supine) on the heating pad. Make a longitudinal skin incision in the inguinal area, approximately 15 mm in length, parallel to the linea alba (Fig 1). Use blunt dissection to separate connective tissue (cotton swabs, hemostats, scissors-blunt tip). Retract the incision to fully view the incision area (Femoral artery, vein and nerve).

Using fine tip forceps and cotton swabs separate the femoral artery from the femoral vein. Separation is best when a perpendicular approach is used as this will help avoid vascular tearing.

When bleeding occurs, a sterile cotton swab and/or gauze square should be placed on the segment and pressure should be applied on the area until bleeding ceases, then continue with the surgery.

Place 2 pieces of 4.0 silk suture under the artery individually, such that first thread lies distally towards the leg, while second thread lies towards the body. Tie a loose knot on the side close to the body and place a hemostat in such direction that it creates a tension on the vessel temporarily obstructing the blood flow into the leg. Tie off the distal end suture (triple surgical knot) as far as possible and pull on the silk as far as possible to straighten the vascular access for more direct catheter placement.

Using the micro-dissecting scissors, make a small incision in the femoral artery at a 45 degree angle. Use a small vascular introducer to help insert the 1.6F Pressure Catheter.

Note: Refer to How to Optimize Scisense Pressure & PV Catheter Life Span Technical Note (RPV-200-tn) for suggested Catheter handling techniques.

Insert the Catheter into the vessel though the incision point (straightening the vessel to create tension often aids with insertion). After the Catheter is fully inserted into the femoral artery (approx. 6-7 cm), carefully tighten the anterior ligature around the artery.

Fig. 1: Femoral artery cannulation and subsequent catheterization

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Rat Intracranial Pressure (ICP) Measurement Cont.

Surgical Approach Cont.

Tip: Try to mark 6-7 cm insertion point on the Catheter before inserting

Record the aortic pressure pulse. The abdominal aortic pressure waveform can be recorded as seen in Figure 5.

INTRACRANIAL SPACE CATHETERIZATION

After removing the hair from the rat's scalp follow pre-op techniques as previously described.

Place the rat onto a stereotaxic apparatus by introducing the ear bars into the ear canal while tightening it into place. Make sure that the head is levelled with a ruler and check for a 90° angle between the ruler and the middle of the animal's scalp. Once level, lock the mouth with the anterior mount of the stereotaxic frame (Fig 2).

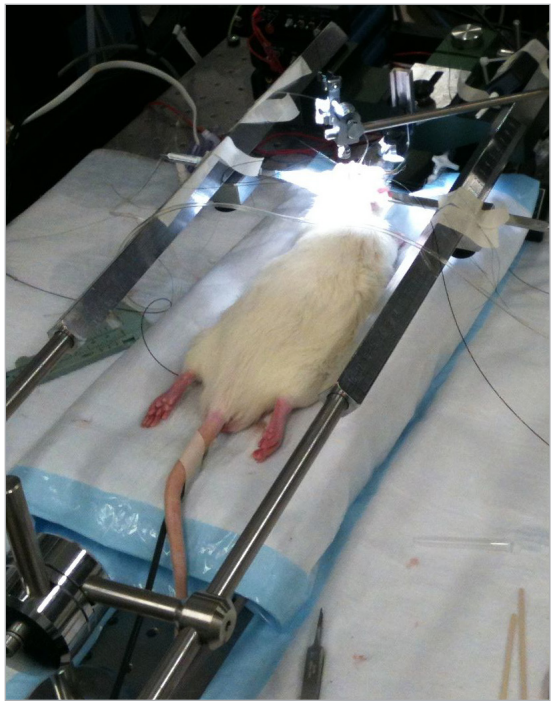

Fig. 2: Rat in stereotaxis apparatus for intracranial surgery

Next, a midline sagittal scalp incision is made to expose the Bregma and Lambda (Fig 3). If necessary, use cotton swabs to dry the exposed skull. The stereotaxic coordinates for intra-parenchymal intracranial pressure monitoring should be used.

The parietal coordinates for the intraparenchymal catheter placement is located 6 mm posterior to the Bregma and 2.5 mm lateral to the midline (Fig 4). Use an Archimedes micro hand drill to fashion a 0.7 mm burr hole over the coordinate. Note: care has to be taken to avoid plunging of the drill bit. Sharply incise the underlying dura mater using the bevel of a 23 gauge needle. The 1.6F Scisense Pressure Catheter is advanced through the burr hole into the brain parenchyma to a depth of 3 mm below the inner table of the parietal bone

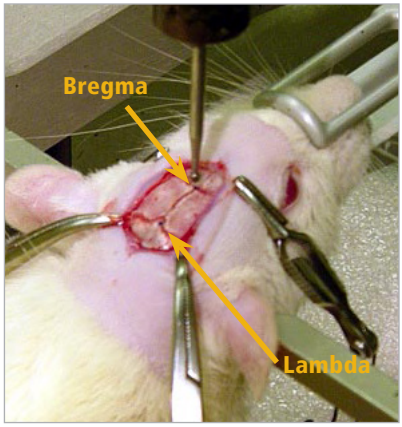

Fig. 3: Incision site for ICP catheterization

At the end of the experiment, carefully remove both Catheters by gently puling it back from the burr hole and from femoral artery (cut the proximal surgical knot before retreating the Pressure Catheter). Immediately, insert the tip of the Catheters into 5 ml syringes filled with saline. Clean Catheters as soon as possible according to proper care guidelines to considerably prolong the Catheter's life (Catheter Cleaning & Disinfecting Guide).

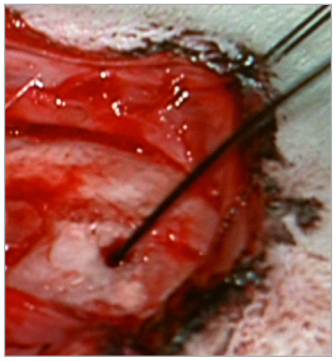

Fig. 4: Inserting Catheter into the parietal parenchymal.

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Rat Intracranial Pressure (ICP) Measurement Cont.

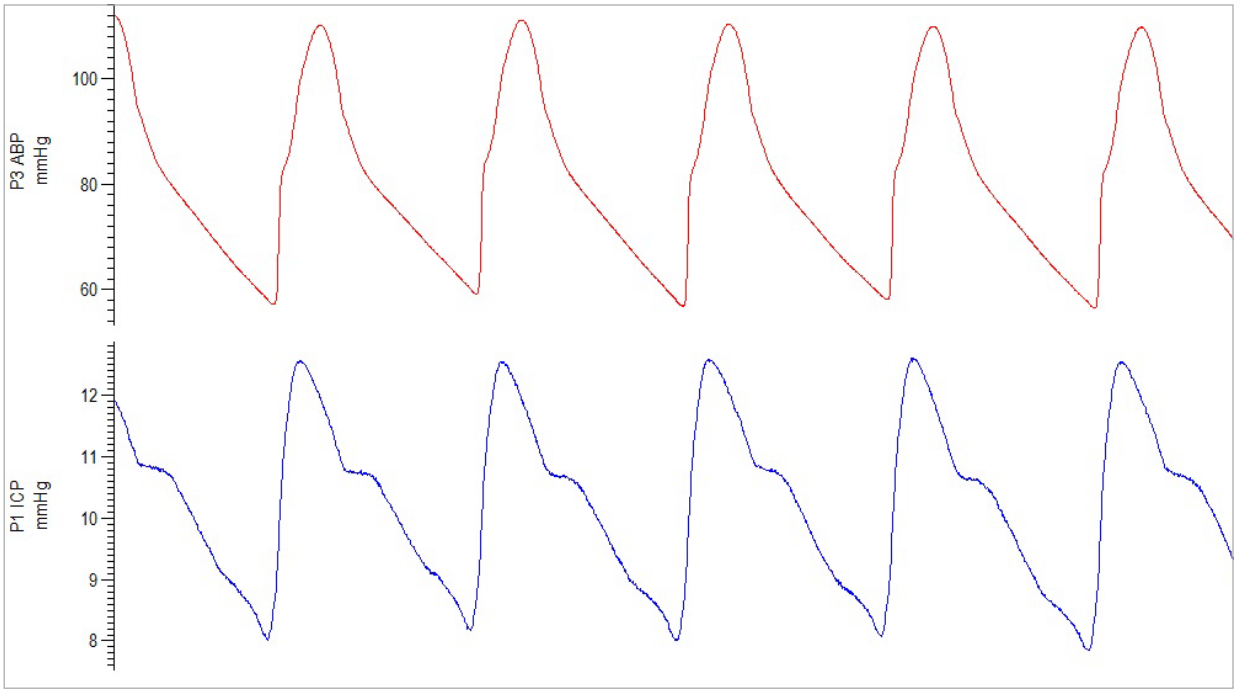

Fig. 5: Representative pressure wave tracings of ABP (arterial blood pressure) in the left femoral artery (red waveform) and Intracranial Pressure (ICP) (blue waveform), obtained by a micro pressure catheter introduced through a burr hole 3 cm below parietal bone.

ACKNOWLEDGMENTS

The animal research protocol used in this study was approved by Macquarie University's animal ethics committee. Methodology, pictures, and supporting data: courtesy of Dr Jonathan Li, Prof Stuart Graham and Prof. Alberto Avolio.

Measurement of Intracranial Pressure (ICP) in Rats© Courtesy of Dr Jonathan Li, Prof Stuart Graham and Prof. Alberto Avolio. The Australian School of Advanced Medicine, 2 Technology Place, Macquarie University NSW 2109, Australia.

REFERENCE

Kim MO, Li J, Qasem A, Graham SL, Avolio AP. Frequency dependent transmission characteristics between arterial blood pressure and intracranial pressure in rats. Conf Proc IEEE Eng Med Biol Soc. 2012; 2012:5614-7.

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