



Oh Rats!

A Guide to Rat Anaesthesia for Veterinary Nurses and Technicians

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Rats are often presented to the clinic to be anaesthetised for minor procedures, routine castration and occasionally for more invasive surgery. Anaesthesia of rats follows the same principles as that of other small animals, however mortality associated with anaesthesia and sedation in rats is much higher than in cats and dogs (Figure 1) (Brodgelt, et al., 2008). The main difficulties associated with rat anaesthesia are lack of familiarity with the species (anatomy and physiology), failure to recognise illness/compromised health, and providing adequate supportive care and monitoring (Thomas & Lerche, 2011). In order to safely anaesthetise rats, the anaesthetist and technician should recognise these difficulties and form an anaesthetic plan to overcome them.

Pre-anaesthetic examination and pre-medication

Rats should be transported to the clinic in a small carry box, although the use of cardboard boxes should be avoided as they may

Species	Anaesthetic mortality (%)
Dog	0.17
Cat	0.24
Rat	2.01
Chinchilla	3.29
Guinea Pig	3.80

Figure 1: Anaesthetic mortality rates for some common household pets (Brodgelt, et al., 2008). This means 2/100 rats die under anaesthesia, versus 2.4/1,000 cats.

chew their way out. Because rats are prey species, carry cages previously used for cats should be avoided as the scent of the cat may cause undue stress to the rat. For the same reason, anyone handling the rat should wash their hands to get rid of the scent of predator species. Rats should also be housed away from cats and other predators if possible.

To restrain a rat it should be picked up around the shoulders and placed on a flat surface, then gently grasped around the shoulders. To hold the rat up for examination or injection it can be picked up by grasping around the shoulders and placing a thumb between the forelimbs and under the mandible to prevent being bitten, with the other hand supporting the hind limbs (Figure 2). It is important not to hold too tightly around the thorax, as this can impair respiration and cause stress and struggling (Longley, 2008).

Prior to anaesthesia, a basic physical examination should be carried out. Stress caused by transport and the physical examination will often mean the heart rate and respiratory rate are markedly increased, however some normal values are included in Figure 3. Like most prey species rats disguise illness; many will not show signs of illness until a disease process is very advanced. Rats also have a shorter lifespan than most common household pets, only two to three and a half years on average. That means a two year old rat may be nearing the end of its natural lifespan and is therefore considered geriatric (Thomas & Lerche, 2011).



Figure 2: Holding a rat for examination or injection (Thomas & Lerche, 2011).

Parameter	Normal Value
Adult body weight	300-500 g
Respiratory rate	70-115 breaths/min
Heart rate	250-300 beats/min
Temperature	37.5-39 °C
Average blood volume	65-70 mL/kg (~30mL total)
Average life span	2-3.5 yrs

Figure 3: Normal physical parameters (Thomas & Lerche, 2011).

Before removing the rat from its cage, normal behaviour such as movement, demeanour, eating and drinking should be assessed. Respiratory rate and character can also be assessed without disturbing the rat's normal respiratory pattern, as they may become stressed when handled. This should be followed by auscultating the lungs with a stethoscope. Since rats are obligate nasal breathers it is imperative to assess the nares for patency before inducing with inhalants. Respiratory disease can be indicated by dyspnoea, increased lung sounds, sneezing, and discharge from the eyes or nose. Rats commonly secrete porphyrin from their eyes and nose in response to stress or chronic respiratory disease, these secretions can look red and be mistaken for a bleeding nose (Thomas & Lerche, 2011).

Heart rate and rhythm can be assessed by auscultating with a stethoscope. The heart rate should be high enough that it is hard to count, less than 200 beats/min is considered bradycardic. Mucous membranes should be pink and capillary refill time should be less than one second. A rectal temperature can be taken, which may also be increased due to stress. A peripheral temperature can be taken by placing a thermometer under the fold of skin in the axilla; adding 1 °C to this measurement gives an estimation of the core body temperature.

Other general signs of malaise in rats include a rough or unkempt coat, soiling around the perineum, sunken eyes, and easily palpable dorsal vertebrae and pelvic bones (Thomas & Lerche, 2011). Any of these signs may warrant abandoning the anaesthesia in favour of further investigation or correction of the problem prior to surgery.

Urine and faecal samples can be easily obtained from rats as they commonly urinate and defecate when handled. Pre-anaesthetic blood work is less common in small mammals due to difficult venipuncture, but some normal ranges are included in Figure 4.

Rats are prone to becoming hypoglycaemic under anaesthesia due to their high metabolic rate and low hepatic glycogen stores, especially if fasted for longer than one to two hours prior to anaesthesia. Rats do not vomit so there is usually no need for fasting before the point of pre-medication, unless it is necessary to reduce the volume of stomach contents for a gastrointestinal surgery (Thomas & Lerche, 2011).

Parameter	Normal Range
PCV	35-55 %
Total protein	55-75 g/L
Blood glucose	3-8 mmol/L
BUN	6-23 mg/dL
ALT	17.5-30 IU

Figure 4: Normal blood parameters (Thomas & Lerche, 2011).

Pre-anaesthetic agents should be strongly considered in rats. There are some distinct goals of pre-medicating before induction of anaesthesia, including sedation, analgesia, chemical restraint, decreased stress for both the patient and staff, and decreased peri-operative drug requirements. Some common pre-medication agents used in rats are:

- **Anticholinergics:** Both atropine and glycopyrrolate can be given to prevent bradycardia by inhibiting vagal response. Cardiac output and blood pressure depend on heart rate in small mammals, therefore maintaining a normal heart rate is important. Some rats have serum atropinesterase, which decreases the duration of action and efficacy of atropine; glycopyrrolate may be a better choice of anticholinergic (Longley, 2008). Atropine can still be used although responses may vary.
- **Phenothiazines:** Acepromazine should tranquilize a rat without immobilizing it.
- **Benzodiazepines:** Diazepam and midazolam cause good sedation and relaxation in rats.
- **Alpha-2 Adrenoreceptor Agonists:** Medetomidine and dexmedetomidine provide sedation and analgesia in rats and should cause immobilization at higher dose rates. Alpha-2 agonists may cause bradycardia.
- **Opioids:** Morphine, buprenorphine and butorphanol can be given to provide pre-emptive analgesia and reduce the amount of inhalant anaesthetic required (Thomas & Lerche, 2011). Opioids may also cause bradycardia.

Pre-medication agents can be administered via subcutaneous or intramuscular route, usually into the scruff of the neck or the quadriceps muscle, respectively (Longley, 2008).

Induction and maintenance of anaesthesia

There are a few practical issues to take into account when inducing anaesthesia in rats.

Because of the difficulty in obtaining intravenous access, induction using this route is rare. Other options include intramuscular and intraperitoneal injection, or inhalation agents. Intraperitoneal administration is thought to be less painful than intramuscular injection, but care must be taken not to pierce the bladder, caecum or liver (Thomas & Lerche, 2011). Some anaesthetic agents are also irritating when given intraperitoneally, such as Ketamine (due to its acidity) (Longley, 2008). Induction and maintenance using injectable agents can be advantageous for a procedure requiring access to the head and neck, and avoids environmental contamination. Some disadvantages of injectable induction include difficult administration, pain on injection, narrow safety margins, and larger dose requirements, which can only be titrated to effect by intravenous administration (Longley, 2008).

Due to limitations with intravenous access, induction of anaesthesia by chamber or mask with inhalation agents is common practice. Advantages of using inhalational anaesthesia over injectable agents include ease of induction, ability to quickly change anaesthetic depth, oxygen provided simultaneously, wide safety margins, and rapid recovery from inhalant agents (Longley, 2008). Clear glass induction chambers should be small enough to fill with vapour rapidly and have a scavenge outlet at the top (Figure 5). A mask can also be used; it should be small enough to avoid dead space and should create a good seal over the rat's nose and mouth to prevent anaesthetic gases leaking into the room (Figure 6). Alternatively, a large dog mask big enough to cover the entire rat can be used as a chamber. Halothane, isoflurane or sevoflurane can be used at slightly higher concentrations than those used in cats and dogs, although isoflurane can be irritating to eyes and mucous membranes (Longley, 2008). To reduce irritation and prevent breath holding, the inhalation agent can be gradually introduced, starting with around 0.5% and increasing in 0.25-0.5% increments over a few minutes. Rats have increased alveolar ventilation because of their high metabolic rates, resulting in rapid uptake and excretion of inhalants, however there is usually a brief period of involuntary excitement during chamber/mask induction.

Intubation is possible in rats using a 14-gauge catheter, however it is technically challenging and secretions can easily block

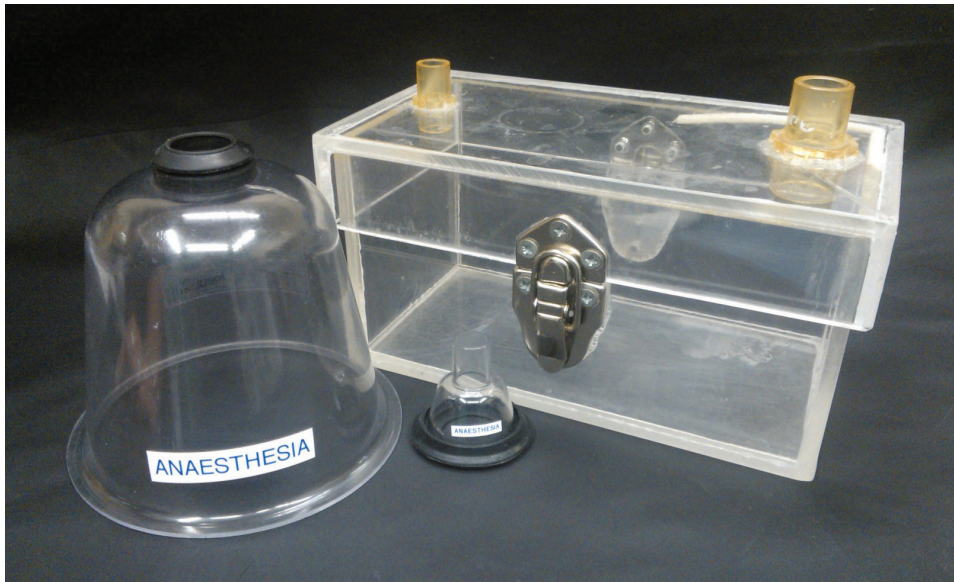


Figure 5: Large mask and small glass chamber suitable for induction of anaesthesia, also showing small mask suitable for a rat.

the small diameter tube. Rats are susceptible to hypoxemia because of their high oxygen consumption rate; therefore it is very dependent on the situation whether the time taken to attempt intubation is justified. Inhalational anaesthesia can be maintained without intubation by using a small mask. Another option is to provide anaesthetic agent and oxygen via a nasal catheter passed toward the back of the pharynx, this may be useful for dental or oral procedures.

Even after mask induction, intravenous catheterisation can be useful to provide

fluids and emergency drugs if required. The lateral tail vein is commonly used, although catheter placement can be difficult in rats. A glove filled with warm water (30–35 °C) can be placed over the tail for a few minutes to cause vasodilation and aid catheterisation with a 24-gauge catheter (Figure 7) (Longley, 2008). Intra-osseous catheters can also be placed but are more technically difficult and have a higher risk of infection.

During anaesthesia rats should be supplemented with dilute (2.5 %) glucose or dextrose in isotonic crystalloid to prevent

hypoglycaemia and dehydration, at a fluid rate of 15 mL/kg/hr. If intravenous access is not possible, 5 mL/kg can be given subcutaneously (provided the solution is dilute). If concentrated, it can cause irritation and sloughing of the skin over the subcutaneous injection site. Fluids should also be warmed as administration of cold fluids can contribute to hypothermia.

Rats have a total blood volume of around 70 mL/kg (~30 mL for an average rat) (Thomas & Lerche, 2011). A loss of 10–30 % of blood volume can be fatal in most species; in rats this is a very small volume of blood, around 3–10 mL, due to their small total blood volume (Longley, 2008). Blood loss can be replaced by intravenous administration of colloids at the same volume of blood lost. Isotonic crystalloids distribute into the extravascular spaces, with only one third remaining in the blood vessels, therefore three times the volume of blood lost must be replaced when using isotonic crystalloids.

Cardiac function can be assessed by monitoring heart rate, mucous membrane colour and capillary refill time. An ECG may not register the small deflections in rats, but a Doppler can be a useful tool for assessing heart rate and rhythm (and therefore give an indication of adequate cardiac output) (Longley, 2008). An area on the ventral thorax between the third and fifth ribs can be clipped and the Doppler probe placed there (Figure 8). Care must be taken not to tape the Doppler probe too tightly around the thorax that it may impair respiration. This does not allow assessment of blood pressure but does provide a constant audible heart rate, therefore arrhythmias and changes in heart rate are easily recognisable. If placed slightly dorsally, the heart beat and respiration can be heard using the Doppler, useful for patients under drapes. The pulse oximeter can be placed on a foot for oxygen saturation, but may not accurately count heart rate (Longley, 2008).

Respiratory function can be assessed by watching the respiratory pattern and rate visually, or using the Doppler to count respiratory rate if the patient is inaccessible under drapes. Typical respiratory rate for an anaesthetised rat can range from 50–100 breaths per minute, anything below 25–50 breaths/min should cause concern (Thomas & Lerche, 2011). A capnograph can still be used in patients that are not intubated



Figure 6: Inducing a rat using a small mask.

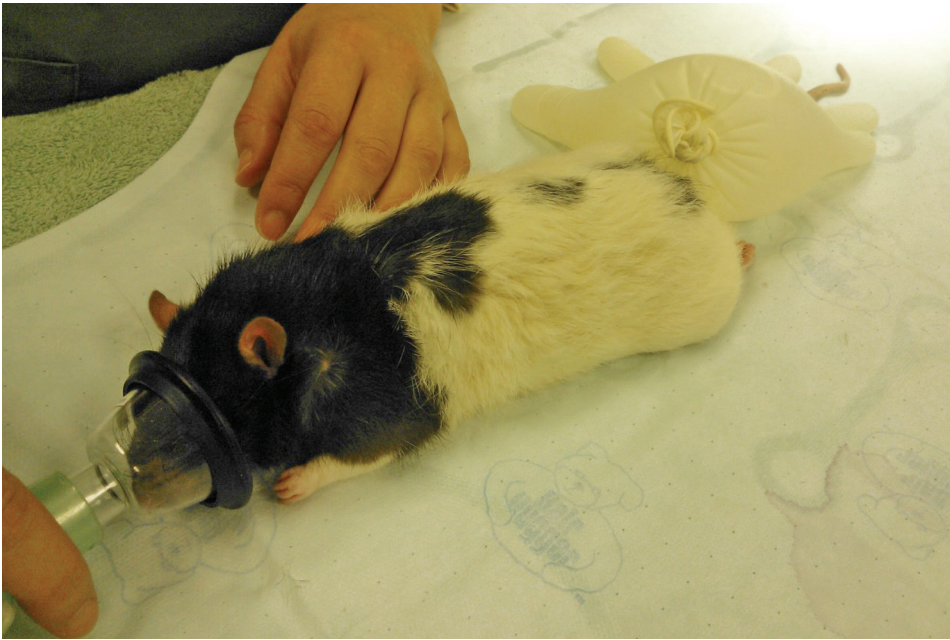


Figure 7: Warm water glove placed over tail to cause vasodilation and aid intravenous catheterisation.



Figure 8: Doppler placement in a rat.

by placing a catheter on the endotracheal tube end of the capnograph line, then feeding the catheter into the mask alongside the patient's nostrils (Figure 9). This may allow for assessment of the respiration rate and an approximation of end tidal carbon dioxide (EtCO₂) levels. The normal range for EtCO₂ in a conscious patient is 35–45 mmHg.

Temperature is an important parameter to monitor in anaesthetised rats; hypothermia can cause bradycardia, arrhythmias, and decreased metabolic rate. Because rats do not have many sweat glands, heat is mostly lost

Figure 9: Capnograph (with catheter feeding towards the nares) and pulse oximeter placement.



through their ears and tail (Longley, 2008). Heat loss can be minimised by keeping a glove filled with warm water over the tail during the procedure (Figure 10). Other ways to prevent heat loss include minimal clipping, using warmed prep solutions, minimising alcohol based solutions, and maintaining a warm ambient temperature. Active heating can be provided during anaesthesia by placing a circulating warm water heat pad under a towel or using a forced warm air blanket under the patient. Wheat packs should be avoided because they can become excessively hot even after short periods of time in the microwave, leading to thermal burns. Hyperthermia should be avoided as heat stress can be fatal in small mammals (Longley, 2008).

Eye reflexes are less useful in rats than in cats and dogs, however pedal withdrawal and tail pinch reflexes are good indicators of anaesthetic depth; in general the tail pinch reflex is lost at a light to medium plane of anaesthesia, followed by loss of the pedal withdrawal reflex at a surgical plane (Thomas & Lerche, 2011).

Recovery and post-operative care

Recovery from inhalational agents is usually rapid compared with recovery from injectable anaesthetic agents. Isoflurane and sevoflurane are mostly excreted by the lungs (i.e. exhaled), with very little (less than 1%) metabolised by the liver. Approximately 20% of halothane is metabolised by the liver. To accelerate recovery some pre-medication



Figure 10: Glove filled with warm water placed over tail to maintain temperature.

agents may be reversed; naloxone is the reversal agent for opioids, flumazenil for benzodiazepines and atipamezole for alpha-2 agonists. Oxygen may be required post-operatively to aid oxygenation, especially in patients that exhibited any degree of respiratory compromise pre-operatively, and may be provided via mask, flow by or oxygen cage. Hypothermia causes shivering, which increases metabolic oxygen demand, therefore hypothermic patients may also require oxygen post-operatively.

Hypothermia is a major complication in recovery due to reduced metabolic rate, meaning slower recoveries as drugs are metabolised and excreted slower (Longley, 2008). Wrapping the rat in a hand towel then wrapping bubble wrap or cling film around the towel will act as insulation and help retain heat (Figure 11). Active warming should be provided until the patient is normothermic or moving around, by the same methods as those used during maintenance of anaesthesia. Take care when providing active warming, especially if the patient cannot move away from the heat source. Heat lamps should be at least one metre away from the patient to avoid thermal burns.

Post-operatively, rats should be encouraged to eat as soon as they are conscious and alert

Figure 11: Rat wrapped in towel and cling film with 100 % oxygen provided via mask.

to prevent hypoglycaemia. They should be offered their normal diet plus extra palatable foods such as warmed baby food (Longley, 2008); if uninterested they may require assisted feeding with a high energy density small mammal feed. Pellets can be soaked in water to increase water consumption (Orr, 2002).

Rats should be monitored for signs of pain and discomfort post-operatively, as pain may decrease appetite and impair successful recovery from anaesthesia (Thomas & Lerche, 2011). Signs of pain include squinting eyes, porphyrin secretion, decreased activity, abdominal writhing, self-trauma, decreased grooming and aggression (Thomas & Lerche, 2011). Provision of adequate analgesia after a painful procedure will aid return to normal function; common analgesics used in rats include buprenorphine, butorphanol,

carprofen, meloxicam and morphine (Longley, 2008).

In conclusion, anaesthesia of rats follows the same principles as that of cats and dogs, with adaptations to suit the patient's size, anatomy and physiology. Nurses and technicians should be familiar with the characteristics of rats in order to recognise any illness or compromised health during the pre-anaesthetic examination. Suitable premedication and induction should be followed by adequate supportive care and monitoring during maintenance of anaesthesia. During recovery, care should be taken to ensure rats are kept warm to prevent hypothermia. Food should be offered as soon as possible to prevent hypoglycaemia and adequate analgesia should be provided. Once the difficulties associated with rat anaesthesia are overcome there should be no reason not to anaesthetise rats routinely in the clinic.

References:

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Case Scenario: 'Walter'

Signalment:

- Sex: Male entire
- Age: 12 months
- Breed: Brown and white hooded rat
- Presented for routine castration

Physical examination:

- Attitude: Bright, alert and responsive
- Temperament: Friendly, curious
- Weight: 520 gm
- Heart rate (HR): 222 beats/min
- Heart rhythm: Strong, regular
- Respiration rate (RR): 200 breaths/min
- Lung sounds: Clear
- Mucous membrane colour (MM): Pink
- Capillary refill time (CRT): <2 seconds
- Temperature: 38.4 °C

Anaesthetic assessment: Fair

Anticipated problems/considerations:

- Hypothermia
- Bradycardia
- Hypoglycaemia
- Haemorrhage
- Hypoventilation
- Aspiration
- Pain

Premedication:

- Butorphanol 0.5 mg/kg IM
- Midazolam 1 mg/kg IM
- Glycopyrrolate 0.01 mg/kg SQ

Induction and maintenance:

- Induction agent: Isoflurane via mask
- Carrier gas: Oxygen
- Circuit: T-piece
- Intubation: Not attempted
- Maintenance: Isoflurane via mask
- Catheter: 24-gauge intravenous catheter placed into lateral tail vein
- Fluids: 2.5 % glucose provided at 15 mL/kg/hr IV
- Monitoring equipment: Doppler over heart, capnograph with catheter feeding into mask, pulse oximeter, rectal thermometer
- Parameters monitored: O₂ L/min, isoflurane %, heart rate, respiration rate, SpO₂, Et CO₂, Et Iso %, mucous membrane colour, capillary refill time, pedal withdrawal reflex, temperature

Complications:

- During anaesthesia, heart rate decreased to 160 beats/min. Isoflurane decreased from 1.75 % to 1.5 %. Additional dose of 0.01mg/kg glycopyrrolate given IV. Heart rate increased to 220 beats/min after around 20 minutes

- During surgery, heart rate increased suddenly to 320 beats/min and patient moved a limb. Surgeon asked to stop, isoflurane increased from 1.75 % to 2 %, intermittent positive pressure ventilation (IPPV) performed until heart rate decreased to 260 beats/min and patient had stopped moving
- Rectal temperature decreased to 36.8°C. Forced warm air blanket turned on to increase temp. By the end of surgery patient temp was 37.6°C

Recovery and post-operative care:

- Isoflurane was turned off and 100 % O₂ provided for six minutes, until patient started moving around
- Patient wrapped in hand towel and cling film
- Food and water were provided once patient had crawled out of hand towel and was moving around cage, looking quiet but alert
- Buprenorphine 0.02mg/kg was administered SQ for analgesia.



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