ANAESTHESIA GUIDELINES FOR AIRWAY MANAGEMENT IN RABBITS

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consider the risks attached to anaesthesia in rabbits and offer advice on the optimum techniques for improving outcomes

RABBITS are commonly presented in practice and most practices will have experience at anaesthetising these patients. However, 1.39 per cent of rabbits die under anaesthesia, with most of these deaths due to cardiorespiratory problems. This compares to an anaesthetic death rate of 0.17 per cent in dogs and 0.00167 per cent in humans (Brodbelt, 2006).

Rabbits undergoing anaesthesia are often assumed to be healthy or having localised disease, but the rabbit may well be in a worse physical state than anticipated and it is worth reviewing the American Society of Anesthesiologists (ASA) physical status classification system, which was developed in 1963 (**Table 1**).

It is important to evaluate where each patient fits on this scale and many of the assumed healthy rabbits may well be a lower grade. Rabbits identified as grade one to grade two have a 0.73 per cent chance of an anaesthetic death, those of grade three to grade five have a 7.37 per cent chance of anaesthetic-related death (Brodbelt, 2006). Most rabbits undergoing anaesthesia are, in reality, grade two to grade four.

TABLE 1. American Society of Anesthesiologists' physical status classification

1. A normal healthy patient.
2. A patient with mild systemic disease.
3. A patient with severe systemic disease.
4. A patient with severe systemic disease that is a constant threat to life.
5. A moribund patient who is not expected to survive without the operation.
6. A declared brain-dead patient whose organs are being removed for donor purposes.

The highest risk period is recovery where 64 per cent of anaesthetic deaths occur, with 40 per cent of these being cardiorespiratory in nature (Brodbelt, 2006).

Rabbits are at higher risk for a variety of reasons.

- They are near obligate nasal breathers and open-mouth breathing is a poor prognostic indicator.
- Nasal and sinus disease is common, leading to respiratory compromise. This is particularly evident if the upper airway is blocked with purulent material, which can be easily identified on CT examination of the head (**Figure 1**).
- Rabbits also suffer from lower respiratory tract disease and cardiac disease.
- They are also very good at hiding underlying pathology and illness.

Medetomidine and ketamine is the most commonly used protocol (59 per cent; Brodbelt, 2006) and rabbits may have the level of sedation deepened with isoflurane or sevoflurane by mask (Brodbelt, 2006; **Figure 2**). However, one of the most commonly reported adverse drug reactions in rabbits (to the VMD in the UK) is due to medetomidine or dexmedetomidine and the effects reported were dyspnoea, tachypnoea, apnoea, bradycardia, cardiac arrest and death (Diesel, 2011). The use of medetomidine or dexmedetomidine did not appear to increase the risk of anaesthetic death over other agents in a survey undertaken in the UK (Brodbelt, 2006).

Measuring effective ventilation is much more important than oxygenation. When breathing 100 per cent oxygen, a pulse oximeter will register at least 97 per cent, which is the situation when breathing room air as well. Thus, for pulse oximetry values to fall, oxygenation has to be appalling when on 100 per cent oxygen (**Figure 3**). Pulse oximetry does not tell us anything about the effective ventilation of the patient. The equipment is also quite insensitive.

An example helps to identify the shortcomings of pulse oximetry. If you had a pulse oximeter attached to your finger and then held your breath it is likely you would become hypercapnic and have to breathe again before the pulse oximeter detected a fall in SPO_2 levels. A capnograph, for example, would have immediately detected the breath-holding and the subsequent hypercapnia as a result. For this reason, the authors' preferred option for anaesthetic monitoring is capnography. Capnography has superseded pulse oximetry and is far more sensitive and "real time".

Capnography and pulse oximetry can be used on conscious patients, for example, to help in the clinical assessment of hypercapnia and hypoxia as mucous membrane colour is unreliable (**Figure 4**). However, no statistically significant relationship is found between SPO_2 and SaO_2, although there is between $ETCO_2$ and $PaCO_2$, but a wide range of $ETCO_2$ values were observed for a given $PaCO_2$ due to hyperventilation and rebreathing.

Capnography is totally reliable under anaesthesia although small dead space connectors and measuring close to the end of the endotracheal tube is important to obtain reliable results (Rich, 1990; **Figure 5**). There is a diffusion gradient of about 5mmHg, which is found in human patients too (Evans, 1977). Capnography does require a secure airway and this can limit its usefulness if the rabbit is not intubated.

Pulse oximetry measures the oxygenation of the patient by the oxygen saturation of the haemoglobin molecules (SpO_2). This correlates well with the arterial oxygen (PaO_2) levels. Normal values should be 97 per cent or more. Caution is to be advised if alpha two agonists are utilised as poor peripheral perfusion can lead to improper functioning of the unit, as can skin pigmentation when it is placed on the ear. Probes can be difficult to attach on the ear in some of the smaller rabbits, but can be attached to the tongue (**Figure 5**) or inserted in the rectum avoiding any issues with pigmentation.

Placing a probe on a shaved digit is another option. Pulse oximetry does not provide information on blood flow of the oxygenation of the rabbit's tissues (which is the important objective). Many machines are manufactured for human use and are unable to detect heart rates faster than 250 beats per minute. Some machines can record rates up to 450 beats per minute and it is very unlikely these rates would be exceeded in rabbits.

Most general practitioners in the UK do not use electronic monitoring for elective rabbit anaesthesia, even if it is present in the practice. The main reason for this appears to be uncertainty associated with interpreting $ETCO_2$ or SPO_2 values. It is of value to routinely set up a monitor on all elective procedures to get nurses and other clinical staff used to normal parameters, before their use in a non-elective procedure, where complication rates are considerably higher (Brodbelt, 2006).

Arterial blood gas analysis should be considered a part of the routine pre-operative and intraoperative evaluation of rabbits (**Figure 6**). This has been shown to be superior to capnography and pulse oximetry in evaluating acid base or electrolyte disturbances, ventilation and oxygenation in a variety of species. Capnography and pulse oximetry are used as non-invasive methods to give near real time results, but lack the accuracy of arterial blood gas analysis. Arterial blood gas analysis is simple to perform in the rabbit.

Arterial blood gas analysis is best used to evaluate the status of the patient over time. Samples are usually taken every 30 minutes to allow for processing and for remedial action to be taken and to evaluate its beneficial effects.

The main limiting factor is not taking the sample, or the cost and time in running a

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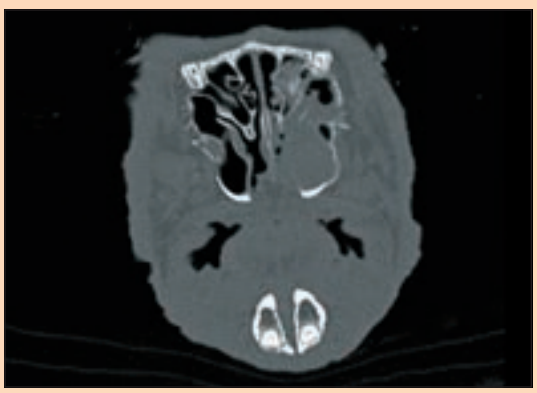

Figure 1. CT scan of a rabbit with a blocked upper airway.

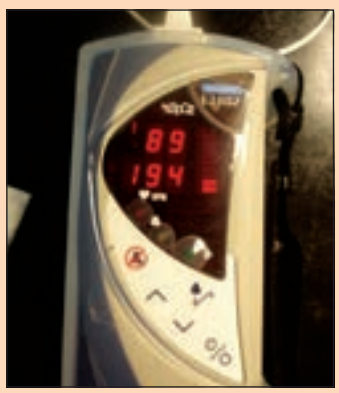

Figure 3 (left). Pulse oximetry reading.

Figure 4 (below). Capnography and pulse oximetry can be used on conscious patients.

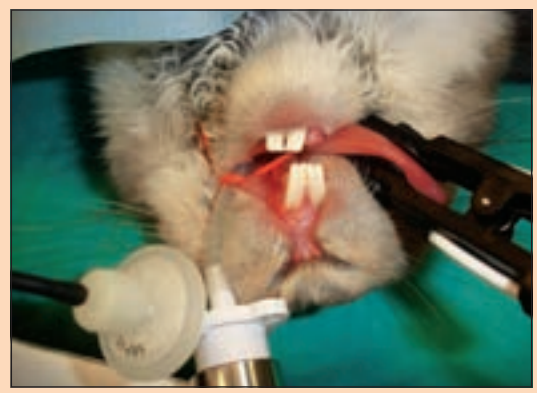

Figure 5. Probes can be attached to the tongue.

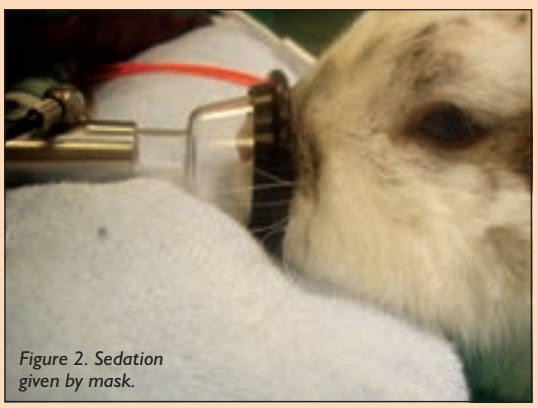

Figure 2. Sedation given by mask.

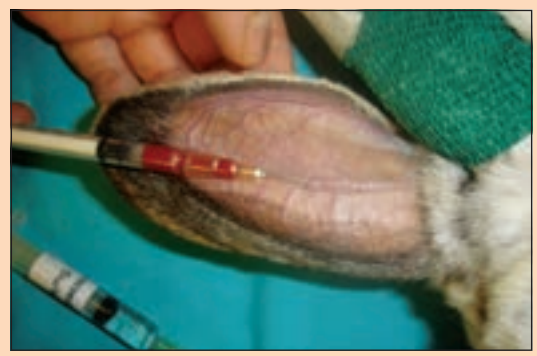

Figure 6. Arterial blood analysis should be part of the pre and intraopertative routine.

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near-patient test, but the initial outlay on a patient-side blood analyser. However, increased availability of these units offered as part of package deals means blood gas and electrolyte analysis can be routine for many patients.

A variety of parameters can be measured, but typically include: pH, PCO_2, PO_2, SO_2, Na, K, Cl, iCal, Glucose, Hct and Hb.

From these, the HCO_3, TCO_2 base excess are typically automatically calculated. The dead space fraction, anion gap and arterial alveolar gradient can be calculated manually.

A number of techniques are employed to assist in the interpretation of blood gas analysis and the necessary remedial action to be taken (Foxhall, 2008).

Mask induction with agents such as isoflurane and sevoflurane has been shown to cause marked breath-holding (up to three minutes) and anxiety, leading to prolonged induction times. As a result, sevoflurane, despite its lower lipid solubility compared to isoflurane, does not lead to quicker inductions in rabbits (Flecknell, 1999).

Should mask induction be performed there is a benefit in pre-oxygenation of the rabbit for a few breaths first, before adding in the anaesthetic gas. One human study showed a marked improvement in oxygenation after four deep breaths in 100 per cent oxygen immediately prior to induction, one to two minutes is therefore excessive and increases anxiety (Fleureaux, 1995).

Wrapping the rabbit in a towel is usually required. This is stressful for any rabbit and vocalisation (screaming) may occur. This breath-holding is still evident after premedication or sedation, although the level of physical restraint required to enable mask induction may be less, and therefore, some premedication is advised as a minimum (Flecknell, 1999). Restraint must be secure as even if mild sedation is used the noxious stimuli from the gaseous agent can still lead to struggling.

In one study, slowly increasing the percentage of anaesthetic gas (every 30 seconds) during induction, did not reduce induction time, nor did it reduce breath-holding, anxiety or struggling, although this is a popular technique. The rabbits held their breath for between 30 and 180 seconds while the lower levels of anaesthetic gas were being administered and started breathing once high concentrations were applied (Flecknell, 1999).

If this technique is to be employed then a potential solution would be to apply a low concentration for the first three minutes of induction, prior to incrementally increasing the concentration. Mask induction with isoflurane is usually at five per cent and sevoflurane eight per cent (due to its higher minimum alveolar concentration). Chamber induction leads to similar anxiety, with rabbits elevating their heads to the top of the chamber avoiding the agent, with subsequent struggling and breath-holding once forced to inhale the anaesthetic gas.

Face masks should be tightly fitting with a membrane over the rabbit's face. Clear masks allow the rabbit to be viewed during induction. There are face masks that allow for some scavenging of waste gases (either active or passive) to reduce environmental contamination. Great care should be taken to ensure the mask membrane does not rest on the corneal surfaces of the eyes. Rubber straps are available that fit over the head to improve the seal. If these are not available, a bandage tie can be placed around the circuit end of the mask and then behind the ears to tie the mask in place. It is also important to prevent the end of the mask from occluding the nostrils if pushed too far on to the face.

Intubation

Only 29 per cent of rabbits are intubated in practice (Brodbelt, 2006). Rabbit intubation can be a tricky procedure and it takes time to master. Face mask anaesthesia has been shown to create clinically significant hypoxaemia and hypercapnia and is best avoided (Bateman, 1995). However, significant tracheal trauma, haemorrhage and oedema can occur with poor intubation technique and respiratory arrest is a potential complication, or protracted failing attempts at intubation (Phaneuf, 2006; Grint, 2006). Many dental procedures are performed without intubation (although it is easy to perform a dental around an endotracheal tube).

The clinician should consider an appropriate airway management technique for each patient. Options involve a face mask for oxygenation or extremely short procedures; a correctly used endotracheal tube; or a supraglottic airway device.

Techniques such as nasal intubation have been described, but should ideally be abandoned once tracheal intubation is mastered (DeValle, 2009). Direct visualisation of the larynx is best because this reduces the risk of tracheal trauma and allows the practitioner to evaluate the oropharynx for food material or foreign bodies. Generally, 2mm to 4mm endotracheal tubes are used. Tracheal trauma is a real risk and repeated attempts to intubate can lead to tracheal and glottal oedema and necrosis (Harkness et al, 2010). This becomes evident usually within a few days post-anaesthesia. All airway devices should be lubricated just before use with a thin layer of a water-based lubricant, which helps to reduce trauma risk.

As with other species prone to upper airway trauma, such as cats, it is essential to provide an adequate depth of anaesthesia prior to intubation or use of a supraglottic airway device. Patients under a very light plane of anaesthesia are more likely to suffer from laryngeal spasm or other upper airway-related complications.

In practice, it is normally sufficient to attain a depth of anaesthesia where applying pressure to the toes or foot webs produces no increase in respiratory rate or any withdrawal reflexes.

Caution and practice is required. Rabbits have a long narrow mouth with a large fleshy tongue and a small gape. This restricted oral access makes intubation difficult. This means it is difficult to see the larynx. They are also obligate nasal breathers and so the soft palate will have to be disengaged from the epiglottis prior to intubation. There are laryngoscopes specifically marketed for rabbits (such as the Flecknell or a Wisconsin blade 0). A long otoscope cone is used as an alternative by many clinicians (**Figure 7**). If metal, the otoscope cone can be autoclaved between patients.

Perfect restraint is needed, although solo intubation is possible with practice. Most rabbits will give a cough response on intubation and many will be quite light by the time they are intubated and some tooth grinding is to be expected. A set time (for example, five minutes) or number of attempts at intubation (such as three) before giving up and resorting to masking the rabbit, will reduce the risk of tracheal trauma, cyanosis and death. Pre-oxygenation before and in between attempts is vital.

The head must be held in a straight line with the rest of the body with the tongue extended and held with forceps. The head is then hyperextended to get the tube over the back of the tongue. The front end of the rabbit may be elevated off the end of the table as well. The otoscope cone can then be inserted via the other side through the diastema.

Other authors prefer to intubate rabbits in dorsal recumbency allowing gravity to facilitate access to the glottis. There are two vascular plexuses either side of the soft palate, which can be visualised easily. The epiglottis can be seen as a v-shaped silhouette behind the soft palate. Flipping the soft palate up with the endotracheal tube will lead to breathing sounds being heard up the otoscope and the glottis can be directly visualised (**Figure 8**). Local anaesthetic (lidocaine) spray can be applied and trickled down the otoscope on to the glottis. The otoscope can be removed and the rabbit oxygenated while this takes effect. Cyanosis is common after the soft palate has been displaced so speed is important, as is oxygenation of the rabbit between attempts.

Some clinicians use an introducer, which is placed via the otoscope into the glottis. The otoscope or laryngoscope is then removed and a premeasured endotracheal tube is passed over the introducer into the glottis. Suitable introducers can include urinary catheters (usually 8-10 French). This reduces the dead space compared to other techniques and reduces the risk of bronchial intubation (**Figure 9**). Others simply pass an endotracheal tube (with the connector removed) without an introducer. A fairly straight endotracheal tube of 2mm to 4mm in diameter will be required. A slight twist of the tube encourages it to fall into place.

Some rabbits may be quite light and the otoscope cone can be used as a mouth gag. A few breaths of inhalant usually allow the rabbit to deepen sufficiently to remove the otoscope cone. Once this is performed the endotracheal tube can be secured in place. Intubation rapidly reverses any cyanosis and capnography can be used to confirm correct placement. Condensation can also be visualised in the endotracheal tube or on a glass slide.

Bronchial intubation is a possible complication (**Figure 10**) and the tube length must be accurately measured. Using an otoscope cone necessitates a longer tube to be used. It must be secured to the head using surgical tape or bandage.

Endoscopic intubation can also be performed. Essentially, the endotracheal tube is passed over the endoscope and then proximal tracheoscopy is performed with the endotracheal tube then slid off the endoscope. A similar technique involves the endoscope being used to visualise the oropharynx allowing accurate endotracheal tube placement.

Blind intubation is also commonly performed. This technique can work well if the veterinarian is skilled in this procedure. Listening to breathing noises compared to gurgling or swallowing noises guides the clinician to where the tube should be placed. However, visualisation is not possible and foreign bodies or trauma are possible. It is also difficult to ensure local anaesthesia has

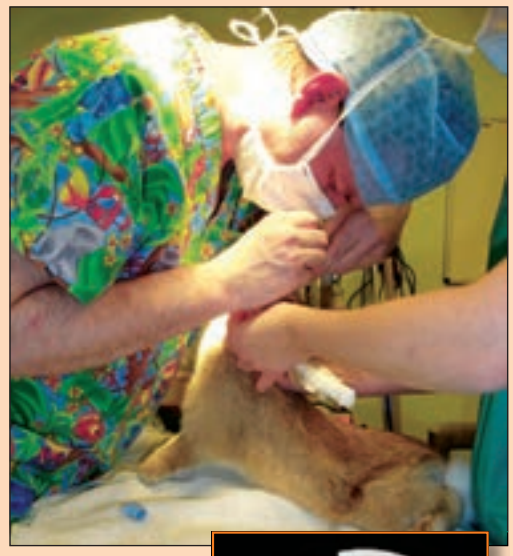

Figure 7 (above). Restricted oral access can make intubation difficult.

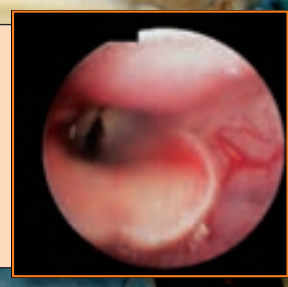

Figure 8 (right). Flipping the soft palate up means the glottis can be directly visualised.

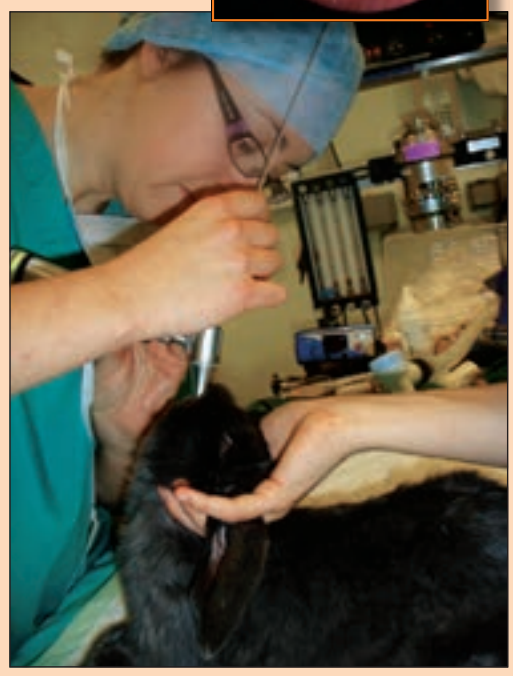

Figure 9. An introducer, such as a urinary catheter, can be placed via the otoscope into the glottis.

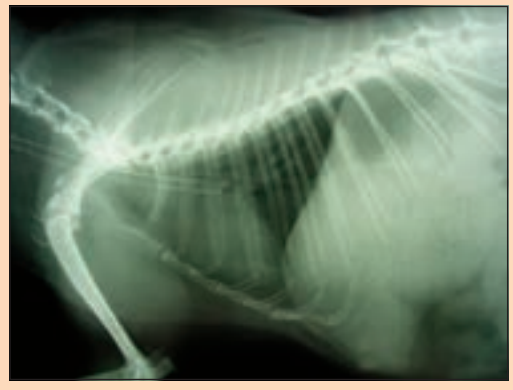

Figure 10. Bronchial intubation can be a complication.

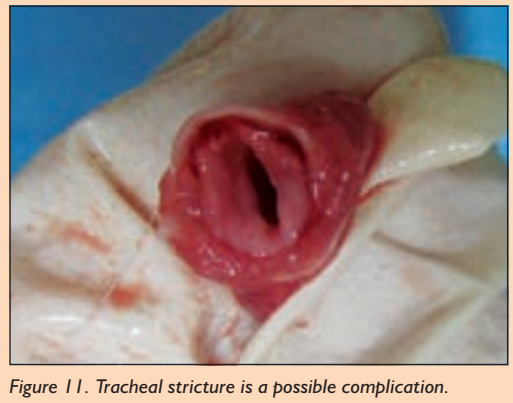

Figure 11. Tracheal stricture is a possible complication.

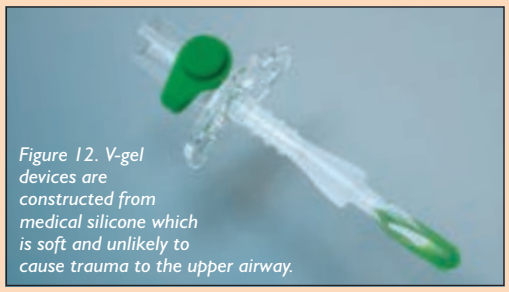

Figure 12. V-gel devices are constructed from medical silicone which is soft and unlikely to cause trauma to the upper airway.

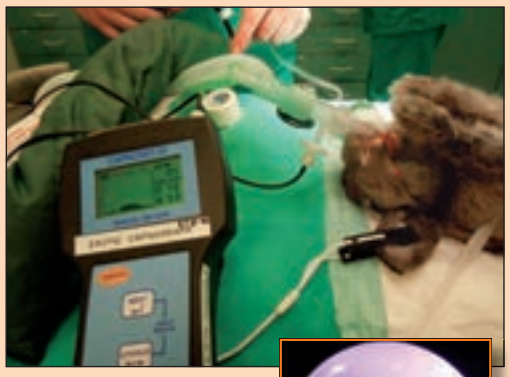

Figure 13. Correct insertion of a v-gel device is best checked using capnography.

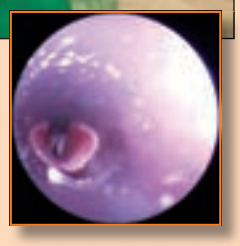

Figure 14. The epiglottis inserts into a v-gel device and unfolds to provide a secure airway.

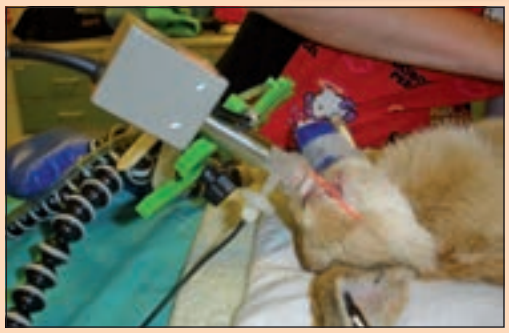

Figure 15. The v-gel device should be secured in place with a restraint.

been applied to the larynx. If this technique is used it is best to use an otoscope to ensure the oropharynx is clear and that the lidocaine spray has actually been applied to the glottis.

Blind intubation of rabbits carries significantly higher risks of upper airway trauma, laryngeal spasm and trauma to the arytenoid cartilages of the larynx. Even if a clinician is skilled in this technique, it is worthwhile to learn one of the techniques allowing direct visualisation of the larynx, as these will reduce the risk of iatrogenic trauma.

Various airway management products are available for rabbits. Although face masks are easy to use, they will leak significant amounts of volatile anaesthetic, increasing staff exposure, will often cause breath-holding and reduce ventilation and give an unreliable airway seal. With training and experience endotracheal tubes can be relatively simple to place.

However, the use of small diameter endotracheal tubes presents another problem. There is an inverse power to the fourth relationship between airway diameter and airway resistance. In other words, halving airway diameter will increase airway resistance by a factor of 16.

Most intubation procedures in a normal rabbit will roughly halve the available airway diameter (2.5mm diameter ETT and an average of 4mm to 5mm internal tracheal diameter.) For rabbit patients, with a small tidal volume and a tendency to underventilate, this is a very significant problem. It can be largely resolved by using a ventilator for all anaesthetics, particularly if capnography is used to evaluate the $ETCO_2$ levels. Most clinicians do not use a ventilator, and airway management methods, such as supraglottic airway devices (see below), which do not narrow the airway, may be more appropriate for this species.

It is essential to properly sterilise any airway device before use to prevent cross-infection with pathogenic oral or nasopharyngeal bacteria. Small bore endotracheal tubes are very difficult to clean properly and great care must be taken to rinse the interior surfaces of the tube perfectly, otherwise airway resistance is increased and there is a risk of inhaling plugs of dried mucus or potentially irritant cleaning chemicals. Tracheal stricture is a possible complication that can occur within 10 days (**Figure 11**).

Plasticisers are chemicals added to plastics during manufacturing and increase the softness of the plastic. Spectrographic analysis of endotracheal tubes demonstrates progressive loss of plasticisers during cleaning with cold sterilising agents (Crotaz, 2012). This will result in material hardening with a consequent increase in trauma risk.

Small bore PVC endotracheal tubes are remarkably cheap. Given the potential for cross-infection, difficulty in correct cleaning and the high iatrogenic trauma risk in rabbits, clinicians should consider these as single-use devices. If this is not an option, the number of uses should be recorded and kept to an absolute minimum.

Endotracheal tubes should be secured to prevent rotation, rostrocaudal movement and accidental removal of the devices. Rotation or reposition of an endotracheal tube tends to increase mucosal trauma at the level of the tube bevel, so every effort should be made to limit this movement. In one laboratory study, simple brushing of the mucosa led to strictures within 14 days with a narrowing of 43 per cent (Steehler et al, 2011).

Once intubated, the tube is taped in place (suturing is also an option for facial surgery). A small filter can be attached to reduce respiratory fluid loss and the rabbit is then connected to the anaesthetic circuit.

Supraglottic airway devices engage into the pharynx and provide an airway without touching the larynx or trachea. They are heavily used in human anaesthesia as they offer easy insertion without traumatising the larynx, so allowing recovery without throat pain, soreness or loss of voice (Keijzer et al, 2009).

Supraglottic devices

Research studies have investigated supraglottic devices designed for human use in various veterinary species. Many are difficult to use or clinically less effective. As supraglottic airway devices work by filling and moulding to the pharynx and perilaryngeal structures, they depend on accurately mimicking the anatomy of the pharynx and upper airway. Some devices have been studied that are specifically designed for species use in veterinary anaesthesia (Bateman et al, 2005; Goldmann et al, 2006).

V-gel devices are veterinary species-specific supraglottic airway devices (**Figure 12**). They are autoclavable to prevent cross-infection between patients and do not require cold sterilising agents to keep them clean. They are constructed from medical silicone, which is extremely soft and unlikely to cause trauma to upper airway anatomical structures.

Rabbit v-gel supraglottic airway devices are designed for easy insertion in a few seconds without the need to visualise the airway. However, it is still essential to check the mouth and pharynx before insertion to ensure no food material is present. Correct insertion is best checked using a capnograph and observing a CO_2 trace (**Figure 13**). They are generally much easier to use than an endotracheal tube, but correct use is still vital to avoid problems. The v-gel device deflects the soft palate dorsally to disengage the epiglottis. The epiglottis then tends to insert into the v-gel device and unfold to provide a secure airway (**Figure 15**).

Capnography is recommended. Firstly, as an indicator of a secure airway and secondly, to identify any epiglottic folding during anaesthesia. This can happen if the head is frequently turned during a dental procedure, for example.

If the epiglottis does fold, a simple repositioning of the v-gel device (gentle rostral then caudal movement) is normally sufficient to regain the airway, readily identified by the CO_2 trace on a capnograph.

The v-gel devices have a tough inner airway protector and it is unlikely a rabbit could bite through them. However, it is recommended to remove them well before full recovery, normally when withdrawal reflexes start to return. Oxygenation should then be maintained using a small face mask.

Lingual cyanosis, probably from lingual venous compression, has been noted during anaesthesia from supraglottic devices. No necrosis or dangerous complications from this have been noted during clinical trial work. If cyanosis is identified, it is sensible to relieve pressure on the tongue by intermittently repositioning the device or using a smaller device if appropriate. Patients should be at a full surgical plane of anaesthesia before attempting to insert the v-gel devices. If they are not, pharyngeal muscular contractions tend to eject the device or make it very hard to establish an airway.

Management and observation of the v-gel airway during the maintenance phase of anaesthesia need to be to a high standard. If the patient is moved without disconnecting the circuit, or the v-gel is accidentally dislodged, this could partially diminish or block the airway. Securing the device in place with a restraint device can be performed (**Figure 15**). Capnography enormously assists identification of these issues, as well as improving the general standard of anaesthesia, and should be considered as a minimum standard for rabbit anaesthetic monitoring. Overall, the soft, anatomical design of rabbit v-gel devices makes them easy to place and allows maintenance of anaesthesia through a low resistance airway that can be easily sterilised after use.

It is impossible to predict if supraglottic airway devices will replace endotracheal intubation, but they certainly provide a superior alternative to face mask anaesthesia, providing arterial blood gas analysis results that have no statistically significant differences from intubated rabbits (Cruz, 2000).

● Please note that some drugs mentioned may not be licensed for use in rabbits.

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