

Practical considerations in anaesthetising exotic species



By Keith Simpson BVSc MRCVS AMIIE(Electronics).

The term exotic species is very broad and can cover anything from a skink to an elephant. For the purposes of this article it will be taken to mean those smaller animals encountered in general practice – small reptiles, small rodents and birds, typically under or around 1kg.

The principles of general anaesthesia apply equally to the exotic species as they do to say dogs and cats. Tidal volumes, oxygen requirements, CO₂ elimination, respiratory rates and body temperature must all be maintained under anaesthesia. One of the most common problems in dealing with anaesthesia in these animals is the support and monitoring of adequate respiration, where respiration includes lung ventilation as well as tissue respiration. Monitoring SpO₂ in such small animals can be quite challenging due to the low pulse volumes and difficulty in probe placement. Capnography offers a viable and arguably superior alternative means of monitoring these animals, although this too has limitations as will be explained.

Often the volatile anaesthetic agents are preferred for induction because of the relative ease of administration and control. However this route also presents problems, in that due to the small size of the patient, relative overdoses are not uncommon and the danger of apnoea or respiratory arrest are increased. Not only that but some patients will simply defy you by not breathing your agent for minutes on end! Depending on the species, anaesthesia may involve intravenous agents first, followed by inhalation agents. One word of caution when using 'gas chambers' for small animals: There is a tendency to run in the gas at 5% until the animal goes to sleep and then fish it out. This is often because the time taken to achieve anaesthesia is otherwise too long. The solution is NOT to run in a higher gas concentration, but to choose a smaller chamber. Otherwise the patient, sensing the gas, reduces or holds its breath until it finally succumbs at which point it will take several full lungs of 5% agent which may then prove devastating in terms of respiratory depression/cardiovascular output. Much better to run in a 1.5-2.0% mixture for slightly longer.

Once your exotic patient is asleep you may or may not choose to ventilate artificially. Often IPPV is not employed because of the extra effort required but the rewards far outweigh that effort.

Why use IPPV?

Oxygen is adequately delivered
Anaesthetic agent is adequately and reliably delivered
Waste gases, particularly CO₂, are reliably eliminated

If you rely on spontaneous breathing the three actions above may not necessarily occur as you would like.

But how to control that IPPV?

IPPV can be done manually but extreme care must be taken to avoid over/under-inflation. IPPV can be done with a ventilator but again care must be taken in choosing an appropriate ventilator. You are not going to be able to adapt your Manley for this task! Volume cycled ventilators should generally be avoided for these small animals because the margin of error in volume delivery is very small. If you determine that your budgie requires 3mls tidal volume and its actual requirement is 2 mls that's a 50% over-inflation. Depending on the compliance of the chest, that may or may not cause a big rise in lung pressure. The same will occur if you temporarily restrict the chest (by leaning on it or just resting your hand on it) - the same volume in a restricted space will again lead to a dramatic rise in pressure. Pressure-cycling ventilators are therefore better suited to these small animals. Choose one that can monitor pressures down to a few cm of water pressure and up to at least 25cm.

But how do you determine the pressure to set?

There is no calculation you can do but nature, thankfully is fairly uniform across the species. Most animals require ventilation pressures of between 5 and 12cm water pressure. This applies across the board – from mice to great danes.

Having obtained a pressure-cycled ventilator how do you connect it to your, often tiny, patient?

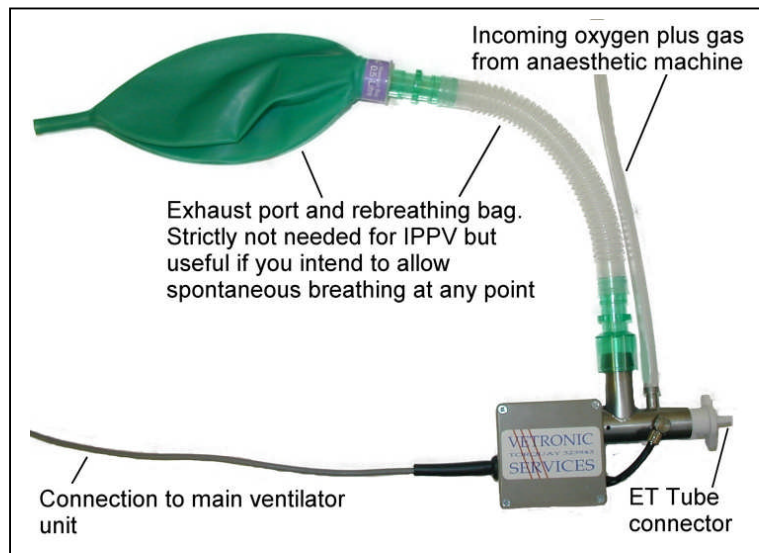
In order to use IPPV your patient is going to have to be intubated. This often requires some ingenuity. Below a few hundred grammes your smallest 2.5mm ET tube is looking far too big. The next step is to use various sizes of intra-venous catheters. You will need to choose one as close to the trachea size as possible. A snug fit will improve your IPPV. These catheters come with a standard luer fitting on them and you will find that this means they will push neatly onto a **2.5mm** Portex ET connector. Also, if you choose one of these connectors with a sampling side port on it, you've got a perfect setup for monitoring end-tidal CO₂ (sidestream – see later) with minimal dead space.

Note - The Portex code for a 2.5mm RSP side port connector is 10025-05S.



Picture 1. Portex ET connector with side-port

The following is a description of the steps to follow when using a pressure-cycled ventilator, based on the Vetronic Services, SAV03 Small Animal Ventilator. This ventilator has been used extensively for exotic anaesthesia since its introduction in 1994.



Picture 2. System connection for IPPV with the Vetronic Services SAV03 ventilator

- Estimate the Minute Volume. See inset for details.

Calculating the Minute Volume

- Weigh your patient
- Calculate the Tidal Volume. A very basic rule of thumb is 10ml/kg tidal volume
- Minute Volume = breaths per minute x tidal volume
- Set the fresh gas flow rate to 3 x Minute Volume
Why 3 x Minute Volume?
There's nothing magical about this, it's just the assumption that the I:E ratio is 1:2. In a spontaneously breathing animal you must provide enough Fresh Gas Flow (FGF) to meet the transient demand of inspiration. With IPPV you want to mimic that. Assuming an Inspiratory to Expiratory ratio of 1:2 you've only got a third of the time to get the gas in, so the FGF must be 3 x the Minute Volume. If the I:E ratio was 1:1 the FGF could be set to 2 x Minute Volume.

- Set the Expiratory length to minimum.
- Set the FGF rate to 3 times the minute volume (see inset).
- Set the trigger pressure to 3 or 4 cm water pressure. You initially won't do much ventilating at this pressure but you won't do any harm either.

- Observe the animal and adjust the trigger pressure for normal chest movements.
- Adjust the Expiratory length to give a normal respiratory rate. Remember that in the very small animals this does not need to equate with their conscious rate. Very small animals expend an awful lot of metabolic energy just breathing, but now you're doing that for them. Therefore, if you keep them warm (a must for any small animal anaesthetic) their oxygen requirement (and CO₂ output) actually drops. Oxygen demand can drop by as much as a third in this situation.
- Adjust the FGF to give a normal Inspiratory duration.

When anaesthetising these patients it is often after gas induction and they may lighten during intubation. This can cause a problem during the initial stages as the animal makes rapid spontaneous breathing efforts. The solution to this is to quickly and adequately ventilate the lungs with gas/anaesthetic and to blow off their CO₂. To do this set the Expiratory time to minimum and increase the Pressure Setting so that the animal receives a full inflation for every breath. This normally overcomes the animals own efforts within about 20-30 seconds.

Having safely anaesthetised and ventilated your patient, how can you assess its vital signs during the anaesthetic?

There are a number of parameters you should be checking routinely, just as you would in any anaesthetic.

Heart rate

Chest movements/respiration rate

Response to noxious stimuli

Body temperature

Just these alone give a good idea of patient status but because respiration is controlled, the only warning sign you may have is heart rate. This may leave you feeling slightly uneasy as respiration may previously have been an important factor in determining anaesthetic depth. A Doppler blood flow monitor can be very reassuring in terms of listening to and following the pulse rate as can a stethoscope. Variations in heart rate may indicate lightening/deepening of anaesthesia, pain response etc. Whilst these are very important, a ventilated patient presents a monitoring challenge as the above do not give a reliable guide to the adequacy or otherwise of the ventilation. Therefore additional monitoring techniques need to be employed. Such additional monitoring aids are

End tidal CO₂

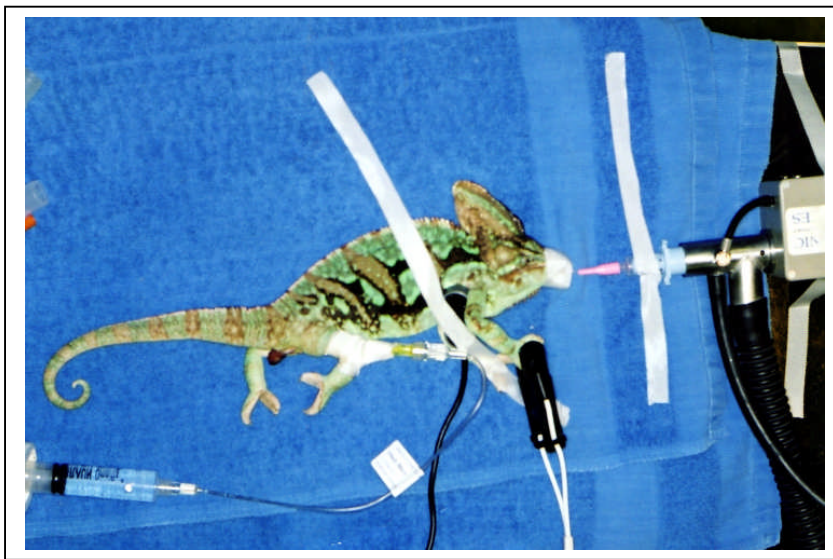
Oxygen saturation

These two can be monitored by a Capnograph and a Pulse-Oximeter respectively. Pulse-Oximetry may not be as useful as anticipated for a number of reasons

- 1) The patients are very small and obtaining a reading can be difficult

- 2) Patients are typically being maintained on 100% oxygen so a severe problem must occur before any fall in SpO₂ is noticed. For animals that are either breathing spontaneously or are being ventilated on room air a Pulse-Ox would be very useful as long as you can get a reliable reading.

If you are going to use a Pulse-Oximeter better readings are likely to be obtained by a reflectance probe rather than a transreflectance probe, although sometimes good readings can be obtained by placing the probe across a foot pad (see Chameleon Photo) or a wing artery. With some small reptiles you can get a reading by placing the reflective probe near the heart. Reflective probes need to be taped into place.



Picture 3.
Intubated
chameleon with
IPPV, intra-
osseous fluids &
SpO₂
monitoring.
Photo Courtesy
of Kevin Eatwell,
Birch Heath
Veterinary
Centre.

Using a Capnograph can give good information on the adequacy of patient respiration. This will almost certainly need to be a sidestream unit if you wish to monitor very small animals. A mainstream unit would require placing in line with a 15mm connector which has two distinct disadvantages

- 1) There is a finite and, depending on the animal, a large dead space volume.
- 2) Your patient needs to be intubated and CO₂ monitoring stops on extubation.

Using a sidestream device will overcome these two problems. A sidestream device draws off a small amount of gas via a sampling tube. This tube can sample from the side-port of an ET connector or directly from a nostril or from inside a face mask. The lower limit of sidestream devices depends upon the sampling rate. The Capnovet-10 from Vetronic Services has a minimum sampling rate of 50ml/minute. This allows animals down to 75 grams to be reliably monitored, although you will see some changes to the waveform in these size animals. A big bonus though is that you can continue to monitor your patient after extubation or monitor animals on a face mask.

There are some human ex-hospital capnograph units on the market which can often be picked up quite cheaply. These will normally have sampling rates of around 200mls/minute which render them unusable for animals below about 2kg so beware of their limitations before you buy.

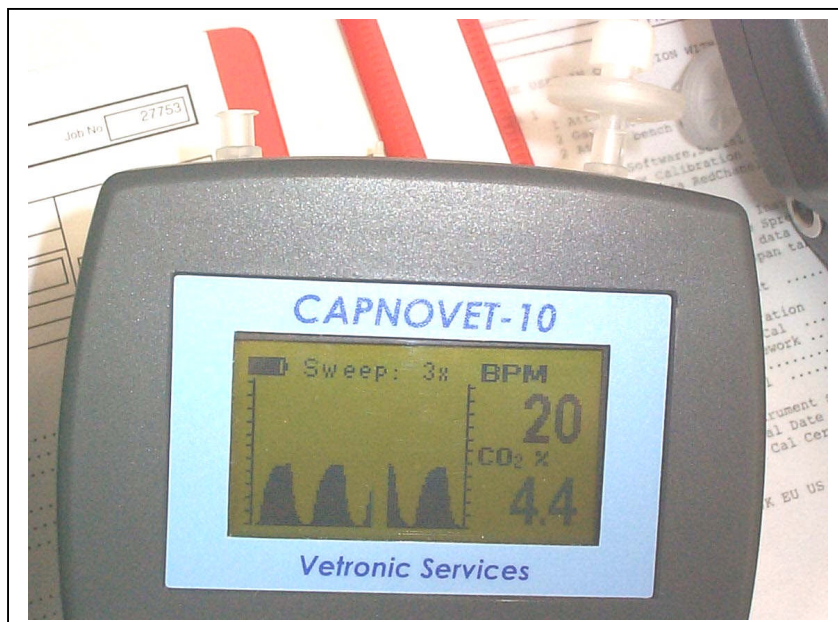
There are many factors that come into play when determining the end-tidal CO₂ value of very small animals. There are unavoidable physical factors of size and anaesthetic set-up etc that will affect not only the end-tidal value but also the waveform appearance. This is equally true of mainstream and sidestream devices. For this reason it is as important to look for trends in CO₂ values and for changes in the waveform profile as it is to look at the absolute values when monitoring these tiny animals. Capnograph units that only have an end-tidal value and no trace are limited in the information they can give. Capnography is extremely useful in any anaesthetised patient but especially so for ventilated animals. Otherwise how do you know how effective your ventilation is?

When using capnography, look for the following:

- Over-ventilation - indicated by a falling End-Tidal CO₂ value.
- Under-ventilation - indicated by a rising End-Tidal CO₂ value.

Aim to keep End-Tidal values between 3.0% and 5.0% (23mmHg and 38 mmHg) to avoid respiratory alkalosis and acidosis respectively.

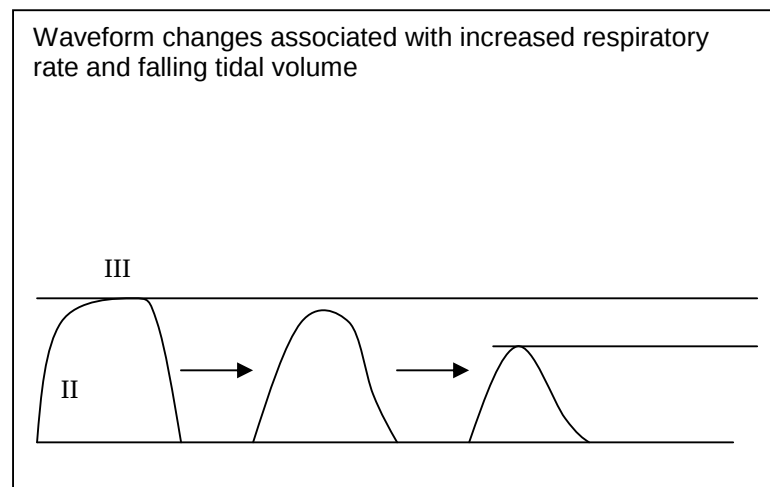
Look for a rapid rise of the wave from baseline on expiration followed by a slowly rising (almost flat) plateau phase. An ideal waveform is shown in photo 4.



Picture 4.
Waveform seen with
ventilated patient.
20 breaths per
minute. End-Tidal
CO₂, 4.4%

As respiratory rate increases the length of the plateau phase will reduce until at rapid breathing rates the waveform looks more like a triangle. A number of factors contribute to this but the dominant factor is the dead-space volume.

Short rapid breaths are associated with a reducing tidal volume until at some point the expiratory volume is approaching the dead space volume. The effect seen then is dilution of expired gas in the dead space. The following diagram shows the effect:



Picture 5. Dilution effects on capnogram appearance

Note the apparent fall in end-tidal CO₂ caused by dilution. As the tidal volume decreases the dilution effect is more marked, hence the decrease in slope of phase II.

The important thing is whether the dead space is predominantly physical (tubing etc), or physiological (just upper airway). If it is predominantly physical the patient may have a normal End-Tidal CO₂ value but a reduced measured End-Tidal CO₂ value. In this instance trying to achieve a plateau phase (no further dilution, constant CO₂ elimination) may prove impossible and you will have to live with the fact that the measured value is less than actual. However, if you are monitoring right at the end of a short ET tube with a small sampling rate on a sidestream unit then the animal has a tidal volume near its physiological dead space. In this situation, with no plateau phase (phase III), the efficiency of CO₂ elimination is reduced and the true End-Tidal CO₂ value may be a little higher or a lot higher than that measured. Only by achieving a plateau phase (no dilution) in this situation can you be sure of the actual End-Tidal CO₂ value.

How to avoid waveforms with no plateau phase

- Use the smallest fittings you can in the common (inspired and expired gas) airway.
- Keep the ET tube length as short as possible.
- Sample for your Capnograph as close to the end of the tracheal tube as possible - use a side-port sampling ET tube connector.
- If not ventilating, then consider IPPV to control the hyperventilation.
- If ventilating and the dead space is mainly physiological, consider increasing the tidal volume and reducing the breathing rate.

Some useful statistics :

Item	Dead Space Volume (max)
ET Connector	1.8 mls
15mm Y-connector	6.0 mls
15mm-15mm connector with oxygen feed	8.0mls
25mm of No.3 ET Tube	0.1mls
Luer hub (female)	0.1mls

Photo 5 shows a rabbit receiving IPPV with carbon dioxide monitoring via an ET connector with side-port.

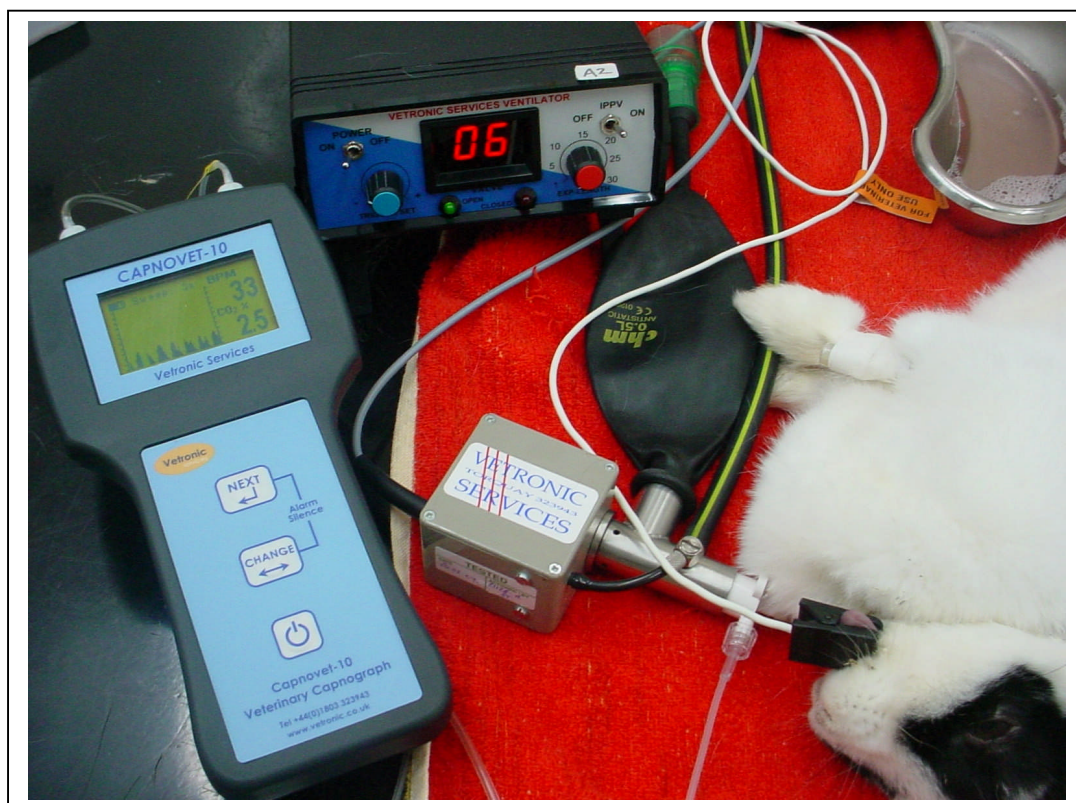


Photo 6. Intubated Rabbit with IPPV, CO₂ and SpO₂ monitoring. Photo Courtesy of Kevin Eatwell, Birch Heath Veterinary Centre.

Summary

Due to the huge array of animal types this article can only be a general guide to anaesthesia in exotic species. Details of intravenous drugs and fluid support are left to other authors. The fundamentals of exotic anaesthesia can be seen to be no different from normal mammalian anaesthesia although, due to their size, assisted ventilation is desirable and with that comes the need to reliably monitor physiological parameters.

Further information on Capnography, Pulse-Oximetry and the Mechanics of Ventilation can be found on the author's web site, www.vetronic.co.uk