1. What do you think are the common pitfalls or are common areas where people go wrong when they look to implement low flow anaesthesia in their practice?

Just to recap that low flow is defined as a fresh gas flow (FGF) of 0.5-1 L/min during the maintenance phase of anaesthesia.

A common problem is animals having too light a depth of anaesthesia, especially after induction of anaesthesia. Even at the start of low flow anaesthesia when we use higher FGF rates (2L/min), this is much lower than those used with non-rebreathing systems (eg Lack or T-piece). So there is a smaller volume of gas carrying the volatile agent. This gas gets diluted in the breathing system by whatever gas is already in the patient’s lungs and in the breathing system (at the start of anaesthesia this is room air). So the vaporiser setting needs to be higher than when using a non-rebreathing system as the volatile gas will be diluted further in the breathing system. This is where measuring inspired and expired volatile agent concentrations can be useful.

Another time when problems can occur is when an animal suddenly becomes light or reacts to stimulation during a procedure. Simply turning up the vaporiser will not deepen the plane of anaesthesia fast enough in low flow anaesthesia. Options are to increase the FGF rate in conjunction with increasing the vaporiser setting; empty the rebreathing bag, which will help to dump the gas with a lower volatile concentration; intermittent positive pressure ventilation (IPPV) and to consider administering a small bolus of the induction agent. It is important to ensure that analgesia is appropriate and additional short-acting opioids such as fentanyl may be appropriate.

On a practical note, I have had problems with leaks in circles. These are often associated with incorrect reassembly after changing soda lime, so always leak test the breathing system before use.
2. How accurate are our vaporisers at lower fresh gas flow rates?

Generally modern vaporisers are considered to be accurate between FGF rates of 0.25 -15 L/min (Boumphrey & Marshall 2011). However, studies have shown vaporiser output to have variable accuracy, depending on the vaporiser type, carrier gases and the FGF rate.

The accuracy of ten isoflurane vaporisers (Isotec 5, Datex-Ohmeda and Sigma Delta, Penlon) using FGF rates of 100% oxygen 1-6 L/min, with vaporiser settings of 0.6-2% was investigated (Kelly & Kong, 2011). The delivered isoflurane concentration differed from the dialled vaporiser concentration by between -50% and +21%. When 100% oxygen was used, the inaccuracy of delivering isoflurane was consistent at all flow rates. However when a 1:2 oxygen:nitrous oxide mix was used, isoflurane was under-delivered at low flows. The study showed that all of the vaporisers tested, at worst were inaccurate to at least ±20%, under-dosing was more likely than over-dosing.

Another study identified that isoflurane vaporisers (Vapor 19.1, North American Draeger) delivered higher concentrations than the vaporiser setting, but at very low FGF rates isoflurane delivery became more accurate (Ambrisko & Klide 2006). The accuracy at different FGF rates will be partly dependent on the make and model of vaporiser and the manufacturer may be able to supply information regarding this. These findings highlight the risks of under or overdosing patients, the benefits of monitoring the concentrations of inspired and expired anaesthetic gases and the importance of carefully monitoring the depth of anaesthesia.
3. What additional advice do you have for those that don’t have access to lots of monitoring equipment?

The ideal monitoring for low flow anaesthesia would include pulse oximetry, capnography (inspired and expired carbon dioxide concentrations) and gas/anaesthetic agent monitoring (inspired O₂, inspired and expired anaesthetic agent concentrations). Gas/anaesthetic agent monitoring is expensive so not commonly used in veterinary practice.

Pulse oximetry is important to give information about the saturation of haemoglobin in arterial blood and this is an essential piece of equipment for monitoring low flow anaesthesia. A damp swab may help improve signal quality when placing the probe on a tongue.

Capnography is useful where available, as it gives information to help assess how well the patient is ventilating. Circle breathing systems have several components (soda lime canister, one-way valves, APL valve, tubing) that can increase resistance to flow and so the work of breathing. While this may not be a problem for the majority of patients, there will be some patients that might not ‘cope’ and so will hypoventilate when breathing through a circle system, or show increased respiratory effort. Hypoventilation can be identified by an increased end tidal carbon dioxide (ETCO₂). Anaesthesia and analgesia drugs will also cause hypoventilation, as will positioning in dorsal recumbency, so the breathing system may be a contributing factor. If hypoventilation is occurring, the severity needs to be assessed, and will depend on the clinical presentation of the patient. Normal ETCO₂ is considered 35-45mmHg, but most healthy dogs can tolerate a higher ETCO₂ up to around 50mmHg (see our capnography resources at https://vetspecialists.co.uk/services/anaesthesia/understanding-capnography/). Manual ventilation is an option to reduce the ETCO₂ back into the normal range. Ventilators can also be used, but we are trying to reduce their use, as oxygen is often used to drive them.

Another use of capnography is to monitor inspired CO₂ levels, which increase gradually when soda lime is exhausted. Soda lime has a dye which changes colour when it is exhausted (from white to purple, or from pink to white). It is important to be familiar with the colour change expected. The problem is that if exhausted soda lime is left in the canister and not used, the granules will revert back to their original colour, giving the impression they are not exhausted. So it is important to change exhausted soda lime as soon as is practical after the case. It would be sensible to change the soda lime when about two thirds of the canister contents has changed colour.

While pulse oximetry is likely to be available and really important for safety under anaesthesia, for low flow anaesthesia monitoring the depth of anaesthesia closely is important. Use a higher FGF (2L/min) and vaporiser setting (1-1.5 x MAC) at the start of anaesthesia and provide some manual ventilation initially until spontaneous ventilation occurs (while avoiding hyperventilation and hypocapnia, which may inhibit spontaneous ventilation). Once the animal is at the desired plane of anaesthesia the FGF rate can be reduced (to 0.5-1 L/min), but keep the vaporiser setting higher than when using non-rebreathing systems (1-1.5 x MAC).
4. Are there any situations where you would avoid using low flow anaesthesia?

One of the benefits of low flow anaesthesia is preservation of temperature in the airways. However, in hyperthermic or pyrexic patients, this is a disadvantage and cold gases may help to lower core temperature. A non-warming bacterial filter could be used in conjunction with higher flow rates in a circle. However, the reaction of carbon dioxide with soda lime produces heat, which is unavoidable.

5. Which kind of patients is it appropriate to use a circle system on?

There are several different types of circle breathing system which vary in the amount of resistance they produce to breathing – this affects the sizes of animals they can be used in. As a general guide, circle breathing systems and low flow anaesthesia should be suitable for patients with a bodyweight of 10kg or more. This is for safety for a few reasons: our guidelines are intended to help people starting out with low flow anaesthesia, with experience and the right equipment circles can be used on smaller patients; there is a variation in the types of circle breathing systems, some will be suitable for smaller patients, while some will not; there is a variation in monitoring, when capnography is used, it is possible to monitor ventilation as described earlier.

Also there are weight ranges in the instructions for the different circle breathing systems. For example, the Humphrey ADE should be used with the canister for dogs over 7kg and without the canister for dogs less than 7kg. However, even without the canister the recommended FGF rates of 70-100 ml/kg/min, are more economical with this system than other non-rebreathing systems when the patient is breathing spontaneously. The Burtons semi-disposable circle is recommended to be used in dogs 10-40kg, whereas Burtons Cyclo-flo is designed to be a lower resistance circuit, and can be used in patients from 7-150kg.

It is important to not only consider weight, but also the body condition and conformation of the patient. An 11kg overweight Pug, with a narrow diameter endotracheal tube, may hypoventilate with a circle breathing system more than a lean 7kg Lhaso Apso.

Lastly consider the tubing: in smaller patients the resistance to breathing can be reduced by using smooth internal bore tubing.
6. ISFM Feline Anaesthetic Guidelines recommend that we use a circle system in cats >4kg – what are your thoughts on this?

The American Association of Feline Practitioners has published feline anaesthesia guidelines (Robertson et al., 2018), which recommend the use of non-rebreathing systems in all cats, but ‘modern circle systems with lightweight plastic rebreathing valves and minimal dead space can also be used safely in cats >3 kg’. The importance of minimising dead space and using paediatric tubing is also highlighted. It is sensible to consider the following: experience, equipment (is the circle low resistance with 15mm smooth internal bore tubing?), monitoring (capnography to monitor hypoventilation), the individual patient (weight, condition score, underlying disease) and expected length of anaesthesia (hypoventilation may worsen with increasing time and hypothermia). Consult the manufacturer’s instructions for the circle breathing system being used as to the intended animal sizes. Personally, I would consider using the Humphrey ADE with the soda lime canister in cats over around 4kg, while monitoring capnography, however, I do not use the Burton’s semi-disposable circles in cats.

7. The guidelines recommend using pulse oximetry to monitor whether oxygen supplementation is required during sedation. Any tips on improving signal such as using a wet swab on the tongue?

Yes, a damp swab on the tongue can improve signal quality with many pulse oximetry probes. Under light sedation, it may not be possible to get a good signal from the tongue due to movement, in this case other sites can be used, such as pinna, digits, vulva or prepuce.
8. How do we assess if the patient requires oxygen supplementation pre-induction? Only pulse oximetry? And what if the patient doesn’t allow?

Pulse oximetry probes are usually placed on the tongue of animals and so this is not usually possible while conscious. Also, if pre-oxygenation is being considered, this suggests the patient has a degree of respiratory or cardiac compromise affecting oxygen delivery to tissues, in which case additional stress (such as attempting to place a pulse oximeter probe) should be avoided.

Pre-oxygenation should be considered in any patient where inadequate oxygen delivery is suspected during or after induction of anaesthesia. Examples include brachycephalic obstructive airway disease, anaemia, cardiac failure and thoracic pathology. On clinical examination, mucous membrane colour, respiratory rate and effort and heart rate will also give indications as to whether additional oxygen would be beneficial. Sedation could be considered to reduce stress while avoiding respiratory depression, which can overall help respiratory compromise.

However, the benefits of pre-oxygenation need to be weighed against any increase in oxygen demand resulting from the stress of administering oxygen. Pre-oxygenation with a mask, compared to flow-by oxygen for 3 minutes produced higher inspired oxygen concentrations (90% compared to 30%) and resulted in a longer time to desaturation of 3 minutes compared to 1 minute (Ambros et al., 2018). So, if a mask is tolerated this is a much more effective way of pre-oxygenating dogs.

9. You recommend the use of cuffed endotracheal tubes. If a practice only has red rubber tubes or uncuffed tubes for cats, would you say they need to purchase new tubes or are ok to continue? Can there be a tutorial on how to correctly inflate a cuff?

Cats are at a greater risk of tracheal damage resulting from overinflating endotracheal tube cuffs, or from physical damage due to traction on the tube (e.g. if the breathing system falls on the floor, or when turning the patient without disconnecting the tube). Over a 10-year period there were 11 claims to the Veterinary Defence Society for ruptured tracheas in cats (no claims in dogs) and in all these cases, red rubber tubes were used for dental procedures (Adshead, 2011). So caution and care is needed with the tube selection and cuff inflation. Red rubber tubes have the disadvantage of being more rigid and have cuffs which are low volume/higher pressure. There are tubes available with better characteristics (softer, high volume/low pressure cuffs) which make them a safer option in cats.

In cats we use ‘Mallinckrodt™ Hi-Contour oral/nasal Tracheal Tube Cuffed’. These have a high volume, low pressure cuff, which is thin-walled and flexible to make intubation easier and minimise the risk of tracheal trauma.

The pressure in the cuff can be measured using a ‘Cuffill Digital Cuff Pressure Syringe’ to ensure the correct inflation pressure, not exceeding 20-30 cmH₂O (Hung et al. 2020).
10. Which monitoring equipment do you use at Davies and what would you recommend for primary care practices?

At Davies, we use mainly ‘Datex S/5’ multiparameter monitors. We also use ‘Vet BP Doppler Kit’ from Burtons for Doppler blood pressure monitoring. The ‘Mindray iMEC 8Vet’ is a good option for monitoring pulse oximetry, capnography, non-invasive blood pressure, ECG and temperature.

11. How often do you find you need to change the soda lime in a circle system?

This will depend on how much carbon dioxide is entering the breathing system, so the size of patient (as this affects the minute volume) and the length of anaesthesia and also the size of the soda lime canister. It is sensible to replace the soda lime when around two thirds of the soda lime has changed colour. If inspired end tidal carbon dioxide increases, this is another indicator to change the soda lime.

12. What is the lowest oxygen flow rate you would feel comfortable using?

Low flow anaesthesia is defined as a FGF of 0.5-1 L/min. Oxygen requirement is only 2-3ml/kg/min but we use significantly higher flow rates for safety and ease of use, and also considering the risk of leaks and the loss of up to 250ml/min with capnography sampling. So with appropriate monitoring, in the maintenance phase of anaesthesia, I would reduce the FGF rate to 0.5 L/min. You should also ensure that the reservoir bag always contains sufficient gas to provide the animal’s tidal volume, and is not ever distended.

References

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