Edite Azevedo

Assunto: FW: Ofício n.º 1007
Anexos: 1.jpg; 2.jpg; 3.jpg; 4.jpg; 5.jpg; ICAM-Humane cat population.pdf; Identification methods for dogs and cats.pdf; IFAW sterilization field manual 2014.pdf; Humane_Dog_Population_Management_Guidance_Portuguese.pdf

De: Maria Pinto Teixeira | Animais de Rua [mailto:mariapteixeira@animaisderua.org]
Enviada: 6 de abril de 2017 17:30
Para: Sofia Lima | Animais de Rua <sofia.lima@animaisderua.org>; Assuntos Parlamentares <assuntosparlamentares@alra.pt>; Miguel Costa <micosta@alra.pt>
Assunto: Re: Ofício n.º 1007

Exmo. Sr.

Na sequência do V. Ofício em epígrafe, somos a apresentar o parecer solicitado, bem como alguma documentação que consideramos relevante para análise do mesmo.

Apresentando os meus melhores cumprimentos, subscrevo-me com a mais elevada estima e consideração,

Maria Pinto Teixeira
Directora Geral
mariapteixeira@animaisderua.org
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Número solidário - Ligue 760 300 161 (€ 0,60 + IVA) e ajude-nos a ajudar!
Receba a nossa newsletter e esteja a par das novidades da Associação Animais de Rua.

---------- Mensagem encaminhada ----------
De: **Assuntos Parlamentares** <assuntosparlamentares@alra.pt>
Data: 23 de março de 2017 às 14:56
Assunto: Ofício n.º 1007
Para: "sofia.lima@animaisderua.org" <sofia.lima@animaisderua.org>

Boa tarde,

Encarrega-me o Senhor Presidente da Comissão de Economia de remeter a V. Exa. o presente ofício.

Com os melhores cumprimentos,

ALRAA

Edgardo Goulart
Assistente Tecnico - Atividade Parlamentar

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animais de rua

1884

Exmo. Senhor Presidente
da Comissão Permanente de Economia
da Assembleia Legislativa
da Região Autónoma dos Açores

Assunto: pedido de parecer sobre o projecto de Decreto Legislativo Regional n.º 5/XI – *"Primeira alteração ao Decreto Legislativo regional n.º 12/2016/A de 8 de Julho que estabelece a proibição de abate de animais de companhia e de animais errantes, na região autónoma dos Açores, bem como medidas de redução e controlo dos mesmos"*

I – Do pedido de parecer

Por meio do V/ Ofício n.º 1007 de 23-03-2017, foi solicitado parecer relativamente ao projecto de Decreto Legislativo Regional em referência

Nesse seguimento, apresentamos o seguinte parecer:

II– Projecto de Decreto Legislativo Regional

Vem o mencionado propor a alteração ao n.º 2 do artigo 16.º do Decreto Legislativo Regional *n.º 12/2016/A de 8 de Julho (doravante mencionado abreviadamente como* Decreto Legislativo Regional), *com vista a passar a ter a seguinte redacção:*

"O disposto nos artigos 3.º e 4.º e no n.º 1 do artigo 11.º entra em vigor a 1 de Janeiro de 2018."

Na sua actual redacção o diploma prevê a sua entrada em vigor no 6.º ano posterior à data de entrada em vigor do Decreto Legislativo Regional, ou seja, em Setembro de 2022.

Ora,

O artigo 3.º do Decreto Legislativo Regional estabelece a proibição do abate de qualquer animal de companhia ou animal errante, isto sem prejuízo das excepções previstas no artigo seguinte, isto é, o artigo 4.º (sublinhado nosso).

E o n.º 1 do artigo 11.º o regime contra-ordenacional aplicável.

Relativamente ao proposto no Projecto Decreto Legislativo Regional acompanha-se o entendimento vertido na proposta aqui em análise, de que se é fundamental que as autarquias tenham tempo para se dotar de centros de recolha modernizados e capazes de dar uma resposta eficaz e ética aos problemas gerados pelo abandono de animais de companhia e pela superpopulação de animais errantes, não é menos importante que a proibição do abate seja implementada num prazo que traduza a urgência da questão, não fazendo desta obrigação um propósito distante no tempo e, por isso, pouco premente.

As políticas públicas de controlo de animais errantes estavam, até recentemente, mais concentradas no combate à disseminação de doenças e aos acidentes provocados pelos animais. A partir de 1990, com a conclusão de que a presença de animais nas ruas se origina principalmente do excesso de nascimentos, as autoridades passaram a dirigir as suas preocupações para a questão da superpopulação e consequente abandono, actuando de forma preventiva, procurando a mais recente legislação ir ao encontro desta última política., pelo fato de serem mais eficientes e humanitárias do que as políticas de captura e abate compulsivo de animais.

Associação Animais de Rua – Esterilização e Protecção de Animais em Risco
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mesmo que se encontrem assintomáticos. Testar todos os animais quanto a todas as possíveis doenças infecto-contagiosas traz custos acrescidos e incomportáveis às câmaras municipais.
Ora, parece-nos que tal situação pode ser ultrapassada, mediante a análise clínica que é efetuada caso a caso pelo médico veterinário responsável, nomeadamente, quando o animal se apresentar sintomático, aí sim ser testado.
Mas veja-se, ainda, que nem todas as doenças infecto-contagiosas impedem que o animal tenha qualidade de vida e/ou são consideradas zoonóticas, nomeadamente a Imunodeficiência Felina (FIV), circunstância em que não se justificaria a eutanásia do animal.
A realização dos testes de diagnóstico para despiste de doenças transmissíveis, como por exemplo a Leucemia Felina (FELV) e a Imunodeficiência Felina (FIV), ou outras que, casuisticamente sejam consideradas importantes pelo médico veterinário municipal sem, no entanto, se estabelecer um critério, como por exemplo, os animais se apresentarem sintomáticos, parece-nos imputar aos municípios um custo excessivo que seria melhor utilizado na esterilização de maior número de animais, erradicando assim as principais formas de contágio destas doenças, que são a reprodução (contágio vertical de mãe para crias, no caso do FELV) e as lutas entre machos inteiros, no caso do FIV (contágio horizontal). A necessidade de testar para FELV e FIV é muito discutível, sobretudo quando os recursos são escassos e a realização dos testes implica, necessariamente, que menos gatos serão esterilizados. Além disso, a precisão dos testes positivos para FELV e FIV é dúbia, com grande número de falsos positivos que necessitam de confirmação laboratorial, cuja demora é incompatível com a manutenção dos animais em confinamento.
Assim, uma vez que por força do disposto na alínea b) do n.º 2 do artigo 4.º a eutanásia de animal de companhia ou de animal errante pode ser praticada caso *"padeça de doença incurável que lhe cause sofrimento e diminuição evidente da sua qualidade de vida"*, parece-nos que mediante a revogação da alínea a) o mesmo fim pode ser alcançado, com um maior rigor, mas desta feita, através da avaliação clínica do médico veterinário.

Decorre do disposto no artigo 6.º, n.ºs 2 e 3 e artigo 9.º, n.º 2 a obrigatoriedade de proceder à identificação electrónica dos animais recolhidos, incluindo os animais silvestres e assilvestrados, nos seguintes termos:

i) Pelas associações zoófilas, legalmente reconhecidas, que procedam à recolha e captura de animais errantes, devendo providenciar pelo seu tratamento médico veterinário, esterilização, encaminhamento para adopção e, quando tal não seja possível, pela devolução dos animais ao seu local de origem, devidamente identificados electronicamente, sendo os felídeos identificados através de corte da parte superior da orelha esquerda, e os canídeos através da colocação de uma coleira empregue especialmente para o efeito;
ii) Os animais recolhidos pelos centros de recolha oficial são obrigatoriamente identificados electronicamente, esterilizados, vacinados e desparasitados;
iii) Por forma a distinguir os animais esterilizados dos animais aptos a esterilização, os felídeos serão marcados através do corte da parte superior da orelha esquerda e os canídeos através de colocação de uma coleira empregue especialmente para o efeito, sendo que ambos deverão ser identificados electronicamente.

Relativamente a esta previsão, que vem estabelecer a obrigatoriedade de identificação electrónica dos animais silvestres e assilvestrados que sejam devolvidos ao local onde se encontravam, parece-nos relevante referir que tais disposições deveriam ser adequadas à sua aplicabilidade prática.
De acordo com o *"Identification Methods for Dogs and Cats"* da *World Society for the Protection of Animals*, a identificação electrónica e as coleiras não constituem métodos ideais de identificação de animais silvestres ou assilvestrados, por razões diferentes. Desde logo pelos custos envolvidos, uma vez a identificação electrónica custa em média 5 euros (considerando o dispositivo de microchip (*transponder*)

animais de rua

A este propósito, veja-se que em 1990, a OMS / WSPA concluíam que, no que respeita à política de captura e extermínio, não haveria nenhuma prova de que a mesma tenha produzido efeitos na redução da densidade populacional canina.

As políticas de captura e abate começaram a ser rejeitadas precisamente com a constatação dos enormes gastos despendidos pelos Estados que adoptaram o método de captura e extermínio, sem qualquer resultado prático para o controle da raiva e outras zoonoses, inaugurando-se, a partir da crítica destas experiências fracassadas, a segunda fase das políticas públicas de controlo das zoonoses e da superpopulação dos animais de companhia abandonados nas ruas, com a elaboração do 8º Relatório do Comité de Especialistas em Raiva da OMS, segundo o qual o método da captura e extermínio deixa de ser considerado eficiente, porque não atua na raiz do problema: a reprodução descontrolada e a ignorância dos detentores dos animais.

Assim, conforme as recomendações decorrentes do 8º Relatório do Comité de Especialistas em Raiva da OMS, para se prevenir o abandono e a consequente superpopulação é necessária a adopção de uma série de medidas preventivas pelos poderes públicos, que poderiam ser resumidas nestas sete linhas de acção: a) controlar a população através da esterilização; b) promover uma alta cobertura vacinal; c) incentivar uma educação ambiental voltada para a posse responsável; d) elaboração de legislação específica; e) controle no comércio de animais; f) identificação e registo dos animais; g) recolhimento selectivo dos animais na rua.

As recomendações da OMS têm produzido importantes efeitos em várias partes do mundo, conforme se percebe através das iniciativas, governamentais e não só, que têm sido tomadas visando promover a consciência para a posse responsável e o bem-estar animal e de que são exemplo, em Portugal, o Decreto-Lei nº 276/2001 de 27/10 e suas alterações, bem como a Lei nº 27/2016, de 23/08 e, mais recentemente, a Lei n.º 8/2017, de 03/03.

Deve o Poder Público implementar políticas públicas que promovam a dignidade e o bem-estar dos animais, desde logo, proibindo o seu abate, privilegiando as acções de vacinação e esterilização em massa, assim como de educação para a posse responsável de animais de companhia, visando que se alcance uma real aplicação das normas ético-ambientais relativas à fauna, sendo que esse actuar do Poder Público deverá dar prioridade aos seguintes aspectos: a) ser eficiente, no sentido de modificar condutas e prevenir o abandono futuro de animais; b) ser humanitário e justo, pois os animais são vítimas da irresponsabilidade dos seus detentores; c) ser da responsabilidade de todos: autoridades, profissionais de saúde, educadores, especialistas em bem-estar animal, organizações não-governamentais e cidadãos em geral.

Em face dos considerandos que antecedem, somos de parecer favorável relativamente no Projecto Decreto Legislativo Regional apresentado pelo Bloco de Esquerda, para que o disposto nos artigos 3.º e 4.º e no n.º 1 do artigo 11.º seja alterado, ou seja, antecipando a entrada em vigor da proibição do abate em vigor a 1 de Janeiro de 2018.

III – Do Decreto Legislativo Regional n.º 12/2016/A de 8 de Julho

Para além da matéria objecto do Projecto de Decreto Legislativo Regional aqui em presença, existem alguns aspectos do Decreto Legislativo Regional n.º 12/2016/A de 8 de Julho, que poderão dificultar a implementação eficaz e eficiente desta nova política de controlo da superpopulação animais e de não abate nos canis municipais. Nesse efeito, cumpre-nos oferecer o seguinte:

O Artigo 4.º, n.º 2, alínea a) estabelece que pode ser praticada a eutanásia de animal de companhia ou de animal errante no caso do animal portador de doença infecto-contagiosa incurável.

Não faz sentido prever indiscriminadamente a eutanásia de animais portadores de doença infecto contagiosa incurável, pois tal pressupõe desde logo que todos os animais recolhidos sejam testados,

e impresso de registo na base de dados) por animal. Atendendo ao número de animais que constituem colónia/matilha esta despesa irá levar a uma redução do número de animais intervencionados, uma vez que os recursos financeiros não são ilimitados, condicionando o programa de controlo de animais errantes. As coleiras acabam por cair facilmente ou serem facilmente removidas e podem ter graves implicações em termos de bem-estar animal. Se não saírem facilmente, podem causar cortes nos membros anteriores, e no limite levar à amputação, quando o animal a tenta retirar e pode levar ao enforcamento se ficar presa. O método de identificação de animais errantes esterilizados recomendado pelo IFAW (páginas 26 e 27 do *Field Manual Veterinary Standards for Dogs & Cats, Surgery and Anesthesia, International Fund for Animal Welfare*) e ICAM (página 27 do *Humane Cat Population Management Guidance, International Companion Animal Management Coalition- WSVA, ARC, RSPCA, HSI, IFAW, WSPA*) é o corte da ponta da orelha. De resto, o corte da ponta da orelha esquerda, ou corte lateral em V no caso dos cães, é o sinal internacional indicativo de animal esterilizado.
Veja-se ainda que, sobretudo no caso dos gatos que compõem as colónias ou nos cães assilvestrados, a maioria dos animais não se deixa capturar duas vezes, o que desde logo inviabiliza a leitura do microchip e, consequentemente, tal obrigatoriedade leva a que tenha de se despender custos desnecessários, que podem ser canalizados para outras acções, nomeadamente um maior número de animais esterilizados.

Estabelece o artigo 9.º, n.º 4 que, se, no prazo de 120 dias a contar da notificação por escrito às associações de protecção animal, o animal em causa não for adoptado poderá ser devolvido à liberdade no seu local de origem ou de captura.
Não destrinça a norma se os animais em causa são animais domesticado e sem capacidade de adaptação e/ou sobrevivência na via pública ou animais silvestres, assilvestrados ou, ainda que domesticados, se encontrem habitualmente inseridos numa colónia de rua ou sob a responsabilidade e supervisão de uma comunidade.
O destino dos animais, designadamente a devolução ao local de origem ou captura, deve acontecer unicamente no caso dos animais silvestres, assilvestrados ou comunitários (gatos ou cães de rua) e não no caso dos animais domesticados, que tenham perdido a capacidade de sobreviver sem a intervenção humana.
No caso dos animais que não sejam silvestres, assilvestrados ou comunitários, poderá até confundir-se essa libertação ou devolução ao local de origem ou captura com a figura legalmente prevista para o abandono (responsabilizado contra-ordenacional e/ou penalmente).
O mesmo não se dirá no caso dos animais silvestres ou assilvestrados que, pelas suas características, não se adaptam ao cativeiro, e que reúnam as condições para sobreviver no seu local de origem ou captura.
Ademais, relativamente ao prazo de permanência nos centros de recolha (de 120 dias), no caso dos animais silvestres, assilvestrados ou comunitários, acontece que esta norma vai contra as boas práticas e o bem-estar animal. O stress associado ao cativeiro destes animais silvestres levará ao desenvolvimento de doenças e seguramente poucos ficarão aptos para o programa. Para além disso, são animais que não se deixam manipular para observação, de difícil higienização e tratamento pelo que a logística de manutenção destes animais em cativeiro se deve restringir aquelas situações para as quais existe justificação médica para a sua permanência.
Nos termos do Artigo 7.º do Decreto-Lei n.º 260/2012 de 12/12, na sua actual redacção, que estabelece os princípios básicos para o bem-estar dos animais é definido que "As condições de detenção e de alojamento para reprodução, criação, manutenção e acomodação dos animais de companhia devem salvaguardar os seus parâmetros de bem-estar animal, nomeadamente nos termos dos artigos seguintes." (n.º 1) e que "Nenhum animal deve ser detido como animal de companhia se não estiverem asseguradas as condições referidas no número anterior ou se não se adaptar ao cativeiro." (n.º 2).

animais de rua

O confinamento prolongado de gatos silvestres ou assilvestrados não deve ser considerado uma opção viável, uma vez que o seu bem-estar fica fortemente comprometido, quando devem os CRO e os abrigos respeitar as suas necessidades comportamentais e fisiológicas.
O confinamento prolongado destes animais, num espaço já de si limitado como é o dos CRO, comprometeria a capacidade de resposta às inúmeras solicitações para intervenção em colónias, a eficácia dos programas e até a necessidade regular de higienização e vazio sanitário.
De acordo com os estudos desenvolvidos pela IFAW (e disponíveis no *Field Manual of Veterinary Standards for Dog & Cat Sterilization Surgery and Anesthesia*), mesmo o stress agudo e de curto prazo afecta profundamente vários mecanismos fisiológicos. A imunossupressão causada pelo stress atrasa o processo de cicatrização, levando a pós-operatórios mais longos e a maior risco de infecções secundárias e desenvolvimento de doenças.
Uma vez capturados e enjaulados, os animais silvestres apresentam-se assustados, confusos e severamente stressados, situação que se agrava com a manipulação humana. Daí que, tipicamente, os gatos silvestres submetidos a programas Captura, Esterilização e Devolução (CED) não se mantenham em confinamento por períodos superiores a setenta e duas horas, incluindo o período de recuperação da cirurgia.
Assim, propõe-se que a redacção do n.º 4 do artigo 9.º poderá acompanhar a disposição prevista no n.º 1 do artigo 3.º da Lei n.º 27/2016, de 23/08, que prevê que "*Os animais acolhidos pelos centros de recolha oficial de animais que não sejam reclamados pelos seus detentores no prazo de 15 dias, a contar da data da sua recolha, presumem-se abandonados e são obrigatoriamente esterilizados e encaminhados para adopção, sem direito a indemnização dos detentores que venham a identificar-se como tal após o prazo previsto.*".
Mais se propõe que seja aditado um número 5, que estabeleça que "*Findo o prazo de reclamação, os animais referidos no número anterior podem, sob parecer obrigatório de médico veterinário ao serviço do município, ser cedidos gratuitamente pelas câmaras municipais ou centros de recolha oficial de animais, quer a pessoas individuais, quer a instituições zoófilas devidamente legalizadas e que provem possuir condições adequadas para o alojamento e maneio dos animais.*"
E ainda, um número 6, que excepcione dos números 4 e 5 que estabeleça "*Que os animais silvestres e assilvestrados que se destinem a ser devolvidos ou recolocados no local de origem ou onde foram capturados, no prazo máximo de 72 horas.*"

Com os meus melhores cumprimentos, subscrevo-me com a mais elevada estima e consideração,

Maria Pinto Teixeira

Maria Pinto Teixeira
Directora Geral

HUMANE CAT POPULATION MANAGEMENT GUIDANCE

International Companion Animal Management Coalition

WSAVA Global Veterinary Development
arc Alliance for Rabies Control
RSPCA
HUMANE SOCIETY INTERNATIONAL
IFAW
WSPA

Executive summary

The International Companion Animal Management Coalition has produced this document to provide government bodies and non-governmental organisations with a detailed resource to support them in their development and implementation of effective and humane programmes to manage cat populations.

Over thousands of years, the relationship between cats and humans has evolved, with an estimated 500 million cats living throughout the world today. The size and make-up of individual cat populations can vary significantly, as can the circumstances and environments in which they are found.

There is no single intervention that will work for all situations where cat populations require some degree of management. An initial assessment and consideration of all potential relevant factors must be made before deciding on the most appropriate programme. What is essential is that the programme is comprehensive and focussed on root causes of the roaming cat population, and not solely on treating the symptoms.

This document examines the five stages of a comprehensive cat population management programme and the elements contained within them.

A The initial data collection and assessment: understanding the problem you are facing by asking the right questions, finding out the relevant information and involving everyone who needs to be involved.

B Analysing and interpreting assessment data with consideration of the influential factors in cat population management: what influences the size and make-up of the cat population and people's desire to control that population?

C The components of a comprehensive cat population programme: based on your specific circumstances and selecting the solutions most appropriate to your situation.

D Designing the intervention: the process that is necessary to create a specific programme suited to your needs.

E Implementation, monitoring and evaluation: applying and keeping the programme on track, and making sure it is effective and achieving its goals.

Throughout this document we will be referencing additional resources that will further aid and support the development of an effective cat population management programme.

Contents

Introduction

The International Companion Animal Management Coalition

The International Companion Animal Management Coalition (ICAM Coalition) is made up of representatives from the World Society for the Protection of Animals (WSPA), the Humane Society International (HSI), the International Fund for Animal Welfare (IFAW), the international arm of the Royal Society for the Prevention of Cruelty to Animals (RSPCA International), the World Small Animal Veterinary Association (WSAVA) and the Alliance for Rabies Control (ARC).

This group was set up to fulfil several objectives, including the sharing of information and ideas on companion animal population dynamics with a view to coordinating and improving member organisations' recommendations and guidance. Each organisation has agreed that it is important to strive to improve our mutual understanding through collaboration. We have a responsibility as funding and advisory bodies to ensure we are offering the most accurate guidance, based on the latest available data and concepts, to those involved with cat population management in the field.

We also believe it is important that we endeavour to be transparent and to document our opinions and philosophy whenever possible. It is to this end that this document has been produced – it represents our recommendations at the time of writing, based on the knowledge we have accrued to date, and will be subject to updates when appropriate. We are aware of the incompleteness of data in this field and will strive both to support the collection of new data and to incorporate it into our future discussions, assessments and guidelines.

Who this guidance is for?

This document is intended for use by government bodies and non-governmental organisations (NGOs) that are involved in cat population management. A similar document, presenting guidelines on the humane management of dog populations, was produced by the ICAM Coalition in November 2007 **(www.icam-coalition.org).**

Ideally, responsibility for cat population management properly resides with local or central government. Animal welfare NGOs should not be required to take on the domestic animal management functions that properly reside with the local authority other than through a contractual agreement, with appropriate funding and resources. However, animal welfare NGOs play a key role in guiding and supporting government strategy, so it is important for such organisations to have an understanding of all the components of a comprehensive strategy. This will enable NGOs to target their support where it can be most effective and to make the best use of limited resources.

Nevertheless, historically, the management of roaming cat populations has come under the remit of NGOs rather than government. Often the management of these populations is regarded as a low priority issue compared with dogs. This is because, for example:

- dog ownership sometimes requires a licence and is therefore more likely to be regulated by a central authority
- dogs present a more obvious public health risk to humans than cats, e.g. the predominance of the dog as a source of rabies in humans, hence the focus of rabies control programmes on the dog
- public nuisance problems are more overt with roaming dogs (barking, fighting, urine and faecal contamination)
- the dog has been associated with humans for much longer than cats, and in many societies a higher value is attributed to it as a companion animal
- dogs are more likely to be pedigree i.e. pure-bred and therefore have some monetary value (either the owner has paid a certain sum to have the dog, or is able to sell the dog for a certain sum) – breeding pedigree dogs may, in some countries, require licensing, while most cats are non-pedigree i.e. of no particular breed, do not have any monetary value and are usually obtained and given away for free
- dogs are often expected to work for humans (e.g. herding or guarding) and may be trained to fulfil this role, whereas cats are not usually regarded as being trainable, and it is their natural behaviour (e.g. hunting of rodents) that benefits humans
- the majority of owned cats are allowed to roam freely – where they go and what they do is not regarded as being within human control
- cats are less domesticated and closer to 'wild-type' than dogs and can therefore, in certain favourable climates, survive in the environment with little or no human intervention or support – this perceived lack of dependence also leads some people to consider them less amenable, suitable for, or deserving of management
- the welfare concerns of dog populations are usually more visible, prompting more motivation to manage them.

The aim of the document

As an animal welfare advocate, the ICAM Coalition believes that when population management is deemed necessary, it is essential that it is achieved in a humane manner and ultimately leads to an improvement in the welfare of the cat population as a whole. As NGOs, we also believe it is important that population management is achieved as efficiently and cost effectively as possible due to limitations on resources and our responsibility to our donors.

The aim of this document is to provide guidance on how to assess cat population management needs and how to decide upon the most effective and resource-efficient approach to managing the population humanely.

We are aware that the status, composition and size of cat populations can vary significantly between and within countries, so there is no single intervention that will work for all situations. Therefore, we strongly advocate the need for initial assessment and consideration of all potential relevant factors before deciding on a programme design. The only concept we consider universal is the need for a comprehensive programme that is focussed on causes and not solely on treating the symptom, namely the roaming cat population.

Why do we need to control cat populations?

The need to manage roaming cat populations arises in many situations, including:

- where the welfare of the cats is compromised (see following section)
- where cats present a public health risk to humans, either through the transmission of zoonotic disease (e.g. rabies, toxoplasmosis) or contamination of the environment (through urine, faeces)
- where cats cause a public nuisance e.g. fighting, scavenging for food (at garbage dumps, restaurants, hotels)
- where cats present a risk to other cats through the transmission of disease (e.g. viruses)
- where cats pose a significant threat to wildlife through predation, in particular where this involves endangered species or where a wildlife population is approaching a critical threshold beyond which it cannot recover.

The welfare of roaming cats may be compromised due to a wide range of causes, including:

- high levels of disease
- inadequate food supply, or inappropriate food supply (cats are obligate carnivores, have specific nutrient requirements and cannot survive on a vegetarian or protein- deficient diet)
- lack of shelter and/or extremes of environmental temperature
- high mortality, especially in kittens who are most vulnerable
- road accidents, a common cause of injury and death
- attacks by dogs and other predators
- malicious attacks by humans, including poisoning.

If none of the above situations apply, the welfare of the cats is good and there is no conflict with humans, there may be no indication to intervene and manage a cat population. However, in most cases problems eventually arise in unmanaged roaming cats, resulting in public health issues, nuisance complaints, and animal welfare concerns because of, for example, high cat and kitten mortality.

Where there is a need for management, the ICAM Coalition's aim is to ensure that effective and humane methods are used and to prevent the use of inhumane methods such as:

- cruel methods of catching
- cruel methods of killing (such as poisoning, electrocution, drowning)
- incarceration in poorly equipped or managed holding facilities.

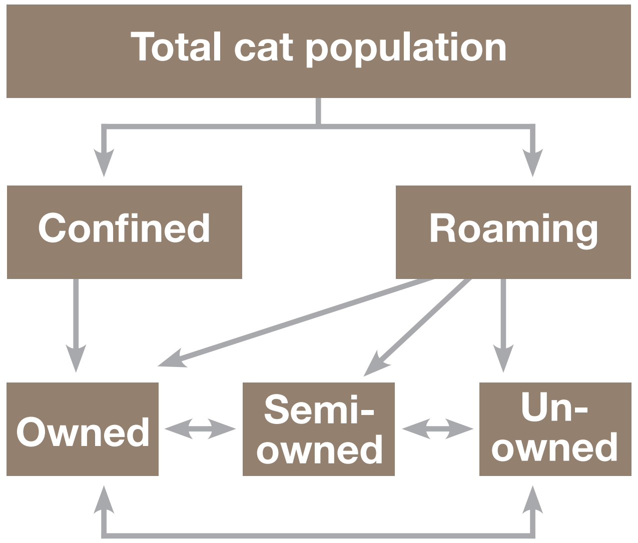
It is relevant when discussing the ICAM Coalition's aim of cat population management to return to an earlier point about the domestication of cats. The process of domestication involves humans imposing a selection pressure for or against certain desired traits. The outcome of this process can be the evolution of a species that fits the human ideal more closely, but a potential side effect is that the species may become less well adapted to an environment without human care. The extent to which this has happened with cats is currently difficult to establish. There is some evidence that cats struggle to maintain good welfare when left without human care from the observation that some cats trapped by trap, neuter, return (TNR) programmes are in a poor state of welfare. However, there is also evidence, not least from some remote islands, where a small founding population has led to the establishment of a large population of cats, that cats can and do flourish without human care. Whether cats are domestic animals that require human care to achieve a good state of welfare or whether they are close enough to their original wild ancestors to be able to survive and maintain a good state of welfare without human care is an important question. This is because the answer would dictate what animal welfare organisations such as the members of the ICAM Coalition would perceive as the desired goal of cat population management.

Should we be aiming for all cats to have adequate human guardianship or is just the absence of persecution enough? In the face of limited data on this subject the ICAM Coalition has taken the decision to err on the side of caution and state as its goal for cat population management to be 'a time when all cats benefit from adequate guardianship'. However, the coalition is interested in exploring this question in more detail and is ready to amend this position in light of more evidence.

Terminology

How to define cats within cat populations can be difficult – the best definitions are those that are workable from a practical aspect. The cat population can be divided into those cats that are confined indoors at all times and those that are free to roam (Bradshaw et al 1999). Cats confined indoors can either be owned or held temporarily in holding facilities such as shelters or foster homes, whereas roaming cats can be owned, semi-owned or un-owned (Figure 1).

Figure 1: Sub-populations of the total cat population
The diagram shows the sub-populations into which the total cat population can be partitioned. These categories are fluid and cats may move between categories, as indicated by the arrows.



Confined cats

Owned cats confined indoors are very likely to be socialised to humans, and have their reproduction controlled by humans. Pedigree cats are more likely to be kept indoors and to have a certain monetary value. Indoor-only cats are unlikely to be a source of unwanted kittens and are unlikely to contribute to the roaming population unless they escape. Except in North America and Australasia, indoor-only cats usually make up a small proportion (less than 20 per cent) of the owned cat population (Rochlitz 2005).

Other confined cats are those kept in facilities such as animal shelters, sanctuaries and foster homes.

Roaming cats

For the purposes of cat population management, it is the roaming cats that are the focus. Within this population, it is most practical to think of roaming cats as belonging to one of three main groups: **owned, semi-owned** and **un-owned,** which are defined below. These definitions are proposed as the most workable for cat population management.

However, because management plans must be adapted to account for the huge variability of conditions where they are necessary, one should be prepared to re-examine the definitions so that they are the most suitable for a particular plan.

1 Owned cats: those for whom an owner can be identified i.e. the person says: "That's my cat".

Owned cats are likely to be owned by an individual, a household, or even a business. An owned cat is a cat that belongs to a specific owner, who cares for the cat by providing food and shelter and who undertakes to be responsible for the cat's welfare. An owned cat is likely to be in reasonable body condition, show some evidence of being socialised to humans and allow itself to be handled (though some owned cats may not). Reliable data are lacking, but a survey in the UK found that the average age of owned cats at death was 12½ years, and the main causes of death were old age/senility, kidney failure, cancer and road traffic accidents (Rochlitz et al 2001). Most cats killed in road accidents were male and less than four years old. These data are likely to be different in other countries.

Most owned cats are allowed outdoors (except in some countries such as the United States of America where approximately 50 per cent are confined indoors or confined in outdoor enclosures), but where they go and what they do when outside is not usually within the control of the owner. When outside, the cat is part of the roaming cat population. Identification of an owned cat can be by microchip, tattoo, or collar, but the majority of owned cats do not have any form of identification. Without identification, it can be difficult to distinguish between owned cats and other categories within the roaming cat population, although owned cats are usually in better physical condition and more socialised to humans.

2 Semi-owned cats: those for whom some kind of caregiver can be identified even if the caregiver does not regard themselves as owners in the conventional sense i.e. the person says "I sometimes feed that cat, or I sometimes offer it shelter, but it does not belong to me".

Semi-owned cats are those living usually in small colonies but sometimes singly, within human communities, on farms, and other locations such as hotels, hospitals and restaurants. They may or may not be socialised to humans. They congregate in these locations because of the availability of food and shelter, which may be provided by a caregiver. A caregiver may be an individual, a household or a business (e.g. hotel staff). Caregivers often enjoy looking after the cats, whose presence is usually appreciated in the community and by local tourists.

It is common for caregivers to feed and provide minimum care for colonies of cats living outdoors because they are concerned about the cats' welfare, but they do not regard themselves as owners of the cats and may be reluctant to take responsibility for them. Caregivers may be prepared to provide these cats with some food and shelter, and possibly emergency veterinary care, but this care does not often extend to providing other aspects of conventional pet ownership such as identification, sterilisation, vaccination or parasite control.

Owners of farms, hotels, restaurants, hospitals, abattoirs and other similar locations may want to maintain small groups of roaming cats for rodent control. The cats are often highly valued for their hunting ability and may have been purposefully acquired for the location. They are likely to live exclusively or primarily on the owner's property, which decreases the likelihood of problems with neighbours.

3 Un-owned cats: those for whom an owner or caregiver cannot be identified.

Un-owned cats may have been previously owned or semi-owned, but for one reason or another may have lost their connection with their owner or caregiver, or they may never have had an owner or caregiver. They may or may not be socialised to humans. These cats find their own food through scavenging and hunting, and make use of whatever shelter is available in the environment. They do not benefit from any veterinary care or other human attention but nevertheless are often partly dependent on resources (food and to a lesser extent shelter) from humans, even if these are not deliberately provided for them (for example garbage dumps and fishing ports). These cats are the most vulnerable group within the roaming cat population, as they do not benefit from any kind of guardianship.

Colony

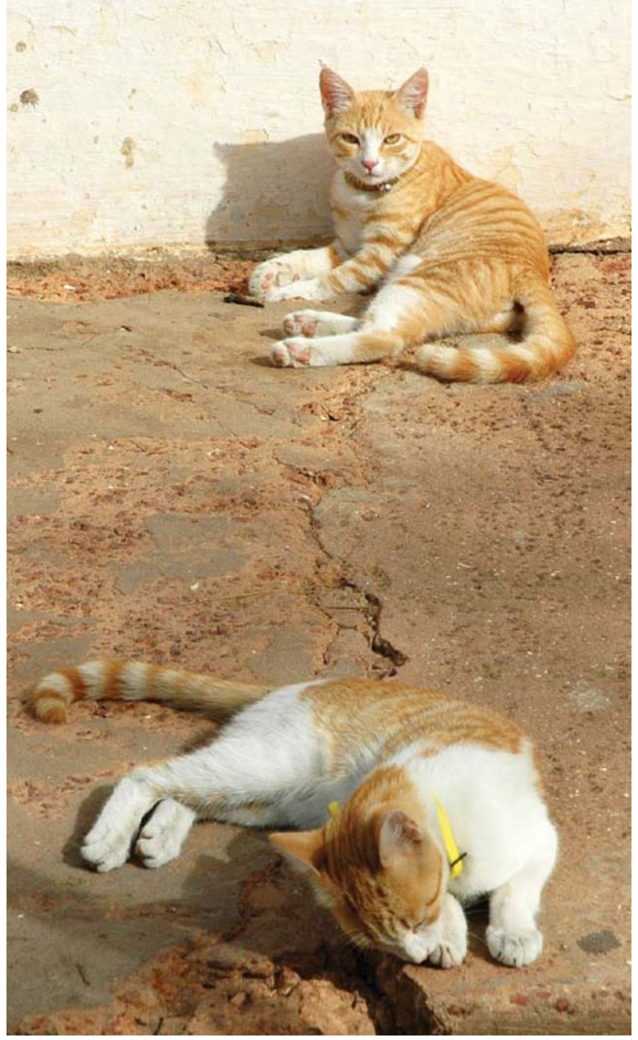
A colony is defined as a group of three or more sexually mature (aged five to six months or more) cats living and feeding in close proximity (Slater 2005). The term is usually used to describe un-owned or semi-owned cats. A managed colony is a group of roaming cats that is controlled by a TNR programme or similar approach (see page 26).

Stray and feral cats

The term 'stray' is sometimes used to describe previously owned cats that have become separated from their owners and are therefore lost, or gone 'astray'. In this case, stray implies that the cats are socialised to humans. Sometimes, however, stray is used to describe all roaming cats, whether socialised or not socialised to humans, and regardless of the cats' sources of food or shelter or ownership status.

While the term 'feral' is often used to describe the un-owned cat that cannot be handled and is un-socialised and therefore not suitable for placement in a home (Slater 2005), the term is also used in a broad sense to include all roaming cats that do not have an identifiable owner, regardless of socialisation status, or the sources of the cats' food or shelter. This includes cats within managed colonies where a caretaker can be identified, other semi-owned cats such as community or barn cats, and un-owned cats that live on the fringes of human communities, such as cats subsisting on rubbish/food dumps. In Australia and New Zealand, the definition of the term 'feral' is closer to the definition of 'feral' used by biologists: cats that live and reproduce in the wild, survive by hunting or scavenging, do not live near centres of human habitation and do not have any of their needs provided by humans.

In this document, in order to avoid confusion the terms 'stray' and 'feral' are not used. Cats are qualified by whether they are owned, semi-owned or un-owned, and if relevant, by whether they are socialised or not socialised to humans. However, when findings from the literature are quoted, the terms used in the source articles are preserved and clarification is provided to be consistent with the above definitions.



Roaming cat populations

The terms free-ranging, free-roaming and roaming are all used in the literature to describe cats that are free to roam in the environment. In this document the term 'roaming' will be used, in accordance with the use of this term in the ICAM Coalition's *Humane dog population management guidance.*

Observations of roaming cat populations

- The distinctions between owned cats allowed outdoors, semi-owned and un-owned cats are often blurred; these cats are all part of the roaming population.
- The population of roaming owned cats is constantly changing. Some owners expect that their cat will go away for a few days and then return. When a cat does not come back, efforts to find it may be limited and often the cat does not have any form of identification. Lost cats are rarely re-united with their owner.
- When owners no longer want their cat, they can easily abandon it somewhere away from their household ('dumping') rather than finding another owner (privately or through a shelter where available). These cats may become less socialised to humans if they do not have contact with them for some time, especially if they were not socialised to humans as kittens during the sensitive period of development (between two and eight weeks of age).
- Some semi-owned or un-owned cats may eventually be adopted and become owned cats, providing they are still socialised to humans.
- The kittens of roaming cats, if they are not socialised to humans by the time they are eight weeks of age, will not usually be suitable as owned cats, but may adapt successfully as semi-owned cats whose needs are met by a caregiver.
- Adult cats that are not socialised to humans are not able to adapt to the existence of a confined owned cat, but may be managed effectively as a semi-owned cat.

Responsible cat ownership

Responsible ownership means that an owner fulfils a duty of care to ensure that their animal's physical and psychological needs are fully met. These include:

- the need for a suitable environment
- the need for a suitable diet
- the need to be able to exhibit normal behaviour patterns
- the need to be housed with, or apart from, other animals
- the need to be protected from pain, suffering, injury and disease.

Instead of focussing on meeting an animal's needs, ownership can also be defined by emphasising the obligations of an owner, i.e. what they should provide. Both cat owners and caregivers should be able to fulfil these obligations.

The minimum obligations of responsible ownership include:

- provision of food, shelter and any other resources (such as social interaction) necessary to maintain an acceptable level of both physical and psychological welfare
- prevention of reproduction (by sterilisation, contraception or other methods unless breeding is planned and homes are available for the offspring)
- identification of cats that have been sterilised by a visual mark e.g. ear tip, tattoo on ear (males and females) or at surgical site (females)
- provision of basic veterinary care (vaccination, parasite control)
- minimising the risks cats may pose to the public or to other animals (in some countries, such as Australia, this is a legal requirement where cat curfews and other restrictions on cat ownership may apply)
- the individual identification of cats – in some countries there may be a legal requirement for owned cats to be identified so that the owner can be traced; however, even if there is no legal requirement, the ICAM Coalition regards this as an essential component of responsible ownership.

In some countries, codes of care are developed to provide minimum and ideal standards of care to ensure owners have guidance on what behaviour is obligated and recommended of them as cat owners. See **www.defra.gov.uk/wildlife-pets/pets/cruelty/documents/cop-cats.pdf** for the UK code of practice for the welfare of cats and **www.biosecurity.govt.nz/animal-welfare/codes/companion-cats** for the New Zealand Animal welfare (companion cats) code of welfare. For 'the five freedoms' see **www.fawc.org.uk/freedoms.htm** for the basic animal welfare framework that underlies many codes of care.

Humane cat population management – a process overview

This flow chart refers to the following sections within this document. It provides a general overview of the stages and processes that need to be considered when embarking on a population management programme.

A. Initial data collection and assessment: understanding the problem you are facing

- Assessing the local cat population
- Data collection methods
- Creating a multi-stakeholder committee

Pages 13 to 15

B. Influential factors in cat population management

- Factors influencing the size of the cat population
- Factors motivating people to control cat populations

Pages 16 to 21

C. Components of a comprehensive cat population programme: selecting the solutions most appropriate to your situation

- Education
- Legislation
- Registration and identification
- Control of reproduction
- Trap, neuter and return (TNR)
- Surgical sterilisation of semi-owned and un-owned cats
- Vaccination and parasite control
- Holding facilities, re-homing centres and foster homes
- Euthanasia of cats
- Control of cat populations using lethal methods
- Controlling access to resources

Pages 22 to 32

D. Designing the intervention: planning, agreeing targets and setting standards

- Aims, objectives and activities
- Planning for sustainability
- Setting standards for animal welfare

Pages 33 to 34

E. Implementation, monitoring and evaluation: checking the programme is achieving its goals

- Implementation
- Monitoring and evaluation

Pages 35 to 36

A. Initial data collection and assessment: understanding the problem you are facing

Before embarking on a cat population management programme it is essential that the dynamics of the cat population are understood and measured objectively. This approach ensures that the final management programme is tailored to the characteristics of the local cat population, instead of being a single blanket intervention for all cats or all situations.

Assessing the local cat population

Initial assessment should include these questions:

- What is the current size of the cat population and what are the categories within it?
- Where are the owned, semi-owned and un-owned cats coming from?
- What are the main welfare issues faced by these cats?
- What is currently being done, both informally and officially, to control the cat population and why?

The fluid nature of the cat population, with many cats disappearing from some households and at least some being added to other households, complicates attempts to measure accurately the dynamics of this population. Ideally, a population estimate should be performed prior to a programme's implementation, but reliably estimating the size of the roaming cat population in a particular location can be difficult. At the very least, a population survey can be used as an indicator of population change over time. In other words, although an estimate of absolute population size may not be that accurate, using the same survey protocol over time would reflect the magnitude of population increase or decrease.

Formal and informal methods of collecting information on the cat populations and on residents' opinions are also crucial to developing the right combination of approaches and obtaining support for population management.

Data collection methods

Household surveys, either door to door or by telephone

Surveys are used to gather data on all aspects of cat ownership, such as: the number of households owning cats; the source of the cat; the cat's sterilisation status; how many litters the cat has had including before sterilisation was done if it was done; whether the household feeds other cats and any welfare problems associated with them and attitudes towards owned cats, the roaming cat population, sterilisation and other issues.

See the following website for examples of household surveys: **www.fanciers.com/npa/studies.html**

Participatory appraisals, focus groups and informal interviews

The aim of focus groups and informal interviews is to explore the subject area from a range of different perspectives, so it is important to ensure that a good representation of the public is included. Interviews are defined as 'informal' when a group of people is asked open-ended questions and the questioning is allowed to develop as a conversation as opposed to following a strict script of pre-defined questions. Participatory appraisals aim to go a step further, by maximising the engagement of local people using flexible and visual tools that don't require people to be literate. This allows local people to guide the direction of the appraisal themselves in order to identify their own priorities for the future programme, rather than just gathering information that someone else uses later to make decisions. The composition of the groups should be carefully considered to ensure everyone feels relaxed and able to respond and discuss the subject matter openly and honestly with the interviewer. Individuals, both cat-owning and non-cat-owning, caregivers and representatives of involved authorities and organisations should be invited to participate in meetings where they discuss all issues to do with the cat population (owned, semi-owned and un-owned), and their attitudes towards them. Representatives of existing shelters, and local groups already involved in cat population control, may have collected data and can be an additional useful source of information.

Indicator counts and mark–resight methods

A population indicator count is simply a count that, under certain assumptions, will indicate whether the number of cats in an area increases or decreases over a period of time. It will not tell you how many cats there are in the area, but a later repeat count can be compared to the original count to indicate if the number of cats has changed.

The method for conducting an indicator count is to select one or more routes across the city or municipality and count cats along those routes. The selected route would need to be recorded accurately so that the count can be repeated consistently. The number of cats seen on a route will certainly be affected by the time of year (especially if there is a clear breeding season), time of day and perhaps by the weather, as well as by the person counting the cats. It is important to try to reduce the effect of these factors by keeping everything the same, as far as possible (i.e. count at the same time of day, avoid times of unusual weather and have the same people involved). It is also necessary to decide on a consistent counting protocol, for example, whether to count cats seen on balconies and roofs or to check for cats under parked cars.

Ideally the indicator counts should be repeated on at least three consecutive days (avoiding any days that may show abnormal cat numbers, for example, due to unusual weather) to find an estimate of how much the counts vary day-to-day. When the indicator counts are compared across years, taking account of changes due to breeding season, any changes in the number of cats can then be compared to the day-to-day variation. If the observed year-to-year change is greater than the day-to-day variation, then it is possible to reject normal day-to-day variation as the reason for the observed change in cat numbers.

The mark-resight method is where cats can be marked (or otherwise identified) and detected later by sighting in order to estimate population size and survival rates. A critical assumption of the mark-resight method to estimate population size is that resighting is independent of marking probability; if cats that are easier to see are also easier to mark this leads to an underestimate; if cats that have been marked become harder to see, this leads to an overestimate.

Once a control programme has started and providing that managed sterilised cats can be marked, for example by having the tip of their ear removed (ear-tipping, see page 27), a mark-resight estimation can be made. This requires good record keeping on the dates of release of marked animals and an estimate of survival. It must be noted that this method of assessment assumes both that marking probability is independent of sighting probability, as explained previously, and that you are working with a closed population. This is a population where there is absolutely no chance of immigration or emigration of animals, especially if the programme that is marking the cats is focused on only a small area within a much larger population of cats. However, as discussed earlier, even if the estimate of population size is not accurate, using the same methodology over time will reflect population size changes over time.

In a survey of a control programme where sterilised cats are marked by ear-tipping, the following equation (Lincoln–Petersen method of analysis) can be used to estimate the total number of adult cats:

Surveys are best conducted at peak cat activity times, but allowing for sufficient daylight, as this is when the most cats will be seen, such as at dawn and dusk. The areas to be surveyed must be clearly defined to avoid straying into areas where cats have not been marked.

Ideally, owners can be asked to identify their cats by collar when a survey is being taken in their locality, so that the population of roaming owned cats can be estimated separately.

Creating a multi-stakeholder committee

Within the perspective of this document, a stakeholder is any person, group of people or organisation that can affect or be affected by cat population management. Creating a committee including representatives from several stakeholders can help improve assessment, analysis and interpretation, design and implementation of a project and finally monitoring and evaluation by benefiting from a range of relevant perspectives. Ideally, it will be the duty of the responsible government authority to bring together stakeholders for consultation. However, NGOs can take the lead in creating a working group that includes the relevant authorities.

The following is a list of possible stakeholders to be consulted. Those marked with * are recommended as minimum requirements of the committee.

- Government * – usually local, but central will also be relevant for policy and statutes and will be the key stakeholder if the programme is national. Several departments are likely to be relevant, including wildlife and conservation, agriculture/veterinary, health, environment (especially with regard to refuse collection), tourism, education and sanitation. (The government must be represented on the committee).
- Veterinary community * – national governing body, veterinary professional association, private practitioner clusters, government vets, and university veterinary department.
- NGO community * – local, national and international organisations working in animal welfare, animal rights, wildlife and conservation, and human health.
- Animal sheltering, fostering and re-homing community * – both government/municipality-run and private/NGO-run organisations.
- Academic communities with relevant experience e.g. in animal behaviour, veterinary science, sociology, wildlife, conservation, ecology and epidemiology.
- Legislators * – departments responsible for both writing and enforcing legislation.
- Educators – in schools and universities.
- Local media – for education, publicity and local support.
- International bodies with relevant responsibilities – such as the World Health Organization (WHO), World Organisation for Animal Health (OIE) and worldwide veterinary associations.
- Local community leaders/representatives *
- Local community – both cat owners and non-owners.

B. Influential factors in cat population management

The data collected during the initial assessment can be analysed and interpreted by taking into consideration two main categories of factors: those that influence the size of the cat population and those that motivate people to control cat populations. Factors that will affect the cat population management plan need to be considered and explored in detail, to ensure that the intervention is suitable for the particular location and circumstances.

Factors influencing the size of the cat population

Reproductive capacity

The sub-population of cats that is contributing most to the population problem needs to be identified, so that management efforts can initially be focused on this sub-population. This could be unplanned litters born from owned cats, as few are sterilised, those born before the owned female cats are eventually sterilised, or litters from semi-owned or un-owned cats, as both these populations are unlikely to be sterilised.

Breeding in cats is not controlled by people to the extent that it is in dogs. Potentially, this is because the majority of cats are not pedigree and of little monetary value or because owners may simply be 'surprised' before they think to control breeding when they encounter the earlier onset and more frequent oestrus cycling in cats as compared to dogs. Compared with dogs, cats appear to be more successful at reproducing and raising a litter to maturity without human intervention or support, as suggested by the existence of islands with cat populations that remain stable or growing without constant recruitment. However, there are no examples of the same situation for dogs.

Pedigree cat breeding is likely to be controlled, with cats and kittens confined indoors, and is probably carried out on a very small scale in developing countries. There is no indication that there is a need for intervention for population control reasons in this small section of the cat population although there may be welfare concerns.

Background information

Cats have a high reproductive capacity. Female cats are seasonally polyoestrus with an anoestrus period associated with day length. Pregnancies can occur throughout the year, but seasonal births are more common and dependent on optimal environmental conditions (i.e. during spring and summer).

Females can have one to two litters a year and one to 10 kittens per litter (Deag et al 2000), with the first litter at five to six months of age. In a survey of US households by New et al (2004), the average litter size (kittens born) was 5.3. There can be high neonatal and juvenile mortality (causes include infectious disease, trauma from road accidents, dog attacks and predator attacks).

In Australia (Toribio et al 2009), the US (Chu et al 2009) and the UK (Murray et al 2009), there are high levels of sterilisation of owned cats (over 80 per cent), but 13-20 per cent of females have mainly unplanned litters before sterilisation.

In a survey of cat ownership and management patterns in central Italy, 43 per cent (39/91) of cats were sterilised, about one in three cats had had a litter, and all litters were considered accidental rather than planned (Slater et al 2008).

1 Roaming owned cats

Owned female cats are likely to receive better health care than semi-owned and un-owned cats, and hence can be an important source of kittens (having more than one litter per year, more kittens per litter and greater kitten survival).

Even if most owned female cats are eventually sterilised, it is often the planned or (mostly) unplanned litters produced by them beforehand that are the most important source of kittens and cats entering the roaming cat population. Owners may want their cat to have kittens before being sterilised, or may be unaware of the age at which cats can get pregnant, or that they can become pregnant again soon after weaning their litter.

2 Roaming semi-owned and un-owned cats

The extent to which these cats contribute to the cat population will vary greatly depending on many factors, including their health status, availability of resources (especially food and suitable nesting sites), number of litters born per year, number of kittens per litter and kitten mortality. The majority of cats are not likely to have been sterilised. Semi-owned cats have a higher reproductive success than un-owned cats as they benefit from at least a minimum level of care (food and shelter provided by a caregiver), but overall their reproductive success is smaller than that of owned cats.

Background information

In an American survey of un-owned (feral) cats, females produced a mean of 1.4 litters per year, with three kittens per litter. Most females were able to produce their first litter at less than 1 year of age. The majority of kittens (75 per cent) died or disappeared by six months of age, and trauma was the most common cause of death (Nutter et al 2004b).

Availability and access to resources

Un-owned and semi-owned roaming cats rely on food and shelter provided intentionally or unintentionally by humans. Food sources include open refuse dumps, household garbage, public bins, as well as households and individuals deliberately feeding cats, in the case of semi-owned cats. Shelter includes structures such as barns, sheds, and garages. The cats' survival and their reproductive success, which includes survival of kittens, often depends on access to these sources of food and shelter, though in some cases un-owned cats can survive and reproduce without human related resources.

Movement of cats between groups within the population

1 Owned cats

Owned cats may become separated from their home, through becoming lost or by deliberately leaving the household, for example, when a male looks for females in oestrus. Conditions in the household may not be suitable for cats so they leave of their own accord. Cats may be returned to their owners if the owners are actively looking for them and/or if they have some kind of identification, but this is uncommon.

They may be deliberately left somewhere away from their home by their owner because they are no longer wanted (abandonment). Abandoned cats reflect a failure of the human-animal bond and are evidence that cats are regarded as disposable and of little value. A cat may not meet the sometimes unrealistic expectations of the owner; owners may also have a poor knowledge of normal cat behaviour.

The option of leaving an unwanted cat at a shelter may not exist, or owners may feel that the welfare of their cat will be better as an un-owned roaming cat than in a shelter.

Background information

Findings from US, UK and Australian data:

- Many cats leave households and many are added to households; 20 to 25 per cent of cats are acquired as previously owned cats that are lost (strays) and are socialised to humans, or are acquired as free gifts from others (Miller et al 1996).
- Over half of cats entering shelters are not relinquished by an owner, but are socialised to humans (Marston and Bennett 2009).
- Reunification of cats with owners is uncommon; few cats (less than five per cent) in shelters are traced back to their owners, as few have any form of identification (Zawistowski et al 1998, Rochlitz 2000).
- Abandonment of cats at sites where population control programmes are in place appears to be a common phenomenon (Levy and Crawford 2004).
- In a study of search and identification methods that owners used to find a lost cat in Ohio, US, the percentage of lost cats recovered by their owners was low, possibly in part because of the lack of use of traditional identification methods (such as a collar) and the general acceptance that cats may roam. Only 19 per cent of cats had some type of identification at the time they were lost. Most cats that were recovered returned home of their own accord or were found in the neighbourhood (Lord et al 2007).
- A survey of 53 animal shelters found that stray cats (those cats that were not relinquished by their owners) entering shelters were much more likely to be re-united with their owners if they were microchipped, although some problems related to microchip registration were encountered (Lord et al 2009).

2 Semi-owned and un-owned cats

Semi-owned cats depend on a caregiver; if this care ceases they become un-owned cats. Un-owned cats do not directly receive care from humans, but some may become semi-owned if they join a managed colony or similar group. If semi-owned or un-owned cats are socialised to humans, they may be adopted (either directly 'from the street' or via shelters or foster homes) and become owned. The movement of cats from owned to semi-owned and un-owned can be substantial, but movement in the other direction is usually less frequent, and depends on how well socialised to humans the cats are and on attitudes of humans towards these cats. Some owners may be reluctant to adopt cats whose previous history is unknown.

Factors that motivate people to control cat populations

Attitudes towards cats

Attitudes need to be explored within households and communities before effective strategies of population control can be devised. If negative attitudes towards cats exist, they will reduce the likelihood of management programmes succeeding, especially if population stability as opposed to reduction is the aim.

Religion and culture play an important role in people's attitudes and beliefs. There may be a belief that sterilisation will cause undesirable behavioural changes, that sterilisation is a form of mutilation or that to deprive an animal of the ability to reproduce is an unacceptable infraction of its rights. Religious and cultural attitudes must be explored and addressed with sensitivity and understanding if they need to be challenged for the benefit of animal welfare.

Surveys from the US show that cats are more likely than dogs to die, be killed, be given away, be relinquished to shelters or be taken away by animal control officers (New et al 2004). More cats than dogs disappear from households, and more are acquired 'off the street'. These findings reflect less attention or concern by cat owners, in general, compared to dog owners, but may not hold true for other countries, for example, see the following background information box.

Background information

A comparison of Bahamian cat and dog caregivers on New Providence JAAWS 12, 30-43 2009 Fielding W.J. Women were more likely to own cats and men more likely to own dogs. Cats were more likely to be adopted and kept as companions than dogs, and more likely to be allowed to live indoors with the family. Cat caregivers appeared to be more attached to their pets than dog caregivers, with dogs being kept as working animals to provide protection. This finding may have been confounded by the fact that more cat caregivers were women, who are reported in the literature as interacting more with cats than men. While many pet owners thought their cats should be sterilised, only a minority of them got their pets sterilised, and cats received limited health care. Despite this, caregivers thought that they were good pet owners.

In this instance, an education programme targeted at women should be developed, which promotes responsible pet ownership and animal welfare, and explores why, despite support for sterilisation, few cats are sterilised. Reasons for not sterilising cats may include cost, the belief that animals have a right to reproduce, concern that the animal's nature will be altered, as well as religious and cultural beliefs. There may also be failure to appreciate the welfare cost to the individual cat, and to the population as a whole, of unregulated reproduction.

Zoonoses

The presence of large numbers of cats, which interact with the human population, raises concern about the transmission of disease from cats to humans. Despite people's fears, the transmission of infections from cats to humans is relatively uncommon. Unfortunately, knowledge within the medical community and public health authorities regarding zoonotic diseases can vary, and veterinary sources may be more reliable.

The following websites present information on zoonotic diseases:

- **www.catvets.com/professionals/guidelines/publications/?Id=181** – A detailed 32-page document on the main zoonotic diseases, how to prevent and treat them.
- **www.capcvet.org/index.html** – A number of articles on the diagnosis, treatment, prevention and control of parasites of clinical importance to dogs, cats and humans
- **www.fabcats.org/cat_group/policy_statements/index.html** – A detailed documents on toxoplasma and cat scratch disease, and three-page summary document on zoonotic diseases and how to avoid infections (rabies not included)
- **www.hpa.org.uk/HPA/Topics/InfectiousDiseases/InfectionsAZ/1191942145653/** – A detailed information on a range of zoonotic diseases affecting cats and humans, from the Health Protection Agency (UK).

1 Rabies

There are a number of countries that are rabies free, but in locations where rabies is present, the following information applies.

Cats acquire their rabies infection from wildlife or dogs, for example, by fighting with a wild animal (mostly raccoons, foxes, or skunks in the US). Hence roaming unvaccinated cats are at the highest risk for rabies infection. The canine rabies variant can also be maintained in unvaccinated dog populations, which in turn may serve as an ongoing source of rabies for both humans and non-humans in a community. However, there is currently no known cat-adapted rabies strain and cats have not been shown to serve as a reservoir of the disease.

Cats will not show signs immediately following exposure to a rabid animal as incubation may be several days, weeks or even months. Symptoms vary, but classic signs of rabies in cats are changes in behaviour, including aggression, restlessness and lethargy, increased vocalisation, loss of appetite, weakness, disorientation, paralysis, seizures and even sudden death.

Rabies vaccines induce a long-lasting immunity, and widespread immunisation campaigns can be very effective. However, legislation to control rabies in cats by vaccination may not exist or may not be enforced in countries where the control of canine rabies is a priority. Nevertheless, all those involved with cat management programmes and roaming cats must be vaccinated against rabies, take all the precautions necessary to avoid being bitten by a cat, and receive prompt wound care and post-exposure treatment if bitten. Sterilised cats may be less likely to encounter infected wildlife because of behavioural changes that result from sterilisation, such as reduced roaming, which may be seen in male cats, especially if they are neutered before puberty (Bradshaw 1992).

Oral vaccination programmes in wildlife can cut the risk of rabies in cat populations by reducing the virus prevalence among wildlife species that might spread the disease to cats. For example see **www.alleycat.org/NetCommunity/Page.aspx?pid=691.**

2 Toxoplasmosis

Toxoplasmosis is caused by infection with *Toxoplasma gondii (T. gondii)*, a coccidian parasite. In people with a normally functioning immune system, toxoplasmosis may be mild and pass undetected or may cause symptoms such as fever and lymph node enlargement.

Toxoplasmosis can cause severe illness in certain 'high risk' groups of individuals whose immunity is impaired. This group comprises:

- developing foetuses
- babies and young children
- very elderly people
- pregnant women (because of the risk to their baby)
- immune-suppressed people.

A cat infected for the first time (typically a young cat) will start to shed millions of oocysts in its faeces after a few days. These oocysts are shed for a short period of time, usually less than 14 days, before the body's immune response stops further shedding. Oocysts shed into the environment do not become infectious until they sporulate, typically after one to five days, so fresh cat faeces do not present a risk.

In most cases, people become infected via one of two routes:

- ingestion of oocysts from the environment e.g. through contact with soil containing sporulated oocysts – this can also occur indirectly through eating soil-contaminated fruit or vegetables
- ingestion of meat containing tissue cysts – fresh meat is most risky since freezing meat for several days or cooking will kill most tissue cysts.

The risks of acquiring toxoplasmosis from a cat are extremely small and most people are infected through other routes (such as eating undercooked meat). Simple everyday hygiene measures can be taken to reduce the risks of infection (from cats and other sources). These include:

- wearing gloves when handling potentially contaminated material (for example, when gardening, cleaning a cat's litter tray or handling raw meat), and making sure to wash one's hands afterwards.
- avoiding eating undercooked meat, and thoroughly washing fruit and vegetables before eating them.

3 Other zoonoses and cat diseases

Other potentially zoonotic infections include plague *(Yersinia pestis)*, cat scratch disease (Bartonellosis), Lyme disease (Borreliosis), *Salmonella, Pasteurella*, roundworms (Toxocariasis) and hookworms, fungal infections (Microsporum, Sporotrichosis), Cryptosporidiosis, and Giardiasis. Fleas and ticks may serve as vectors for cat scratch disease *(Bartonella spp)* and other zoonotic diseases.

Reports of zoonotic diseases in semi-owned and un-owned cats indicate that, for most diseases that have been investigated, these cats do not have a greater rate of infection than roaming owned cats.

In situations where un-owned or semi-owned cat populations are managed through vaccination and parasite control, cats should be in improved health and hence should not pose an increased risk of zoonotic disease or source of infection for other cats as compared with owned cats.

Background information

Nutter et al (2004a) found that feral cats and pet domestic cats had a similar baseline health status and faecal prevalences of infections with Cryptosporidium spp., Giardia spp. and Toxocara cati. Feral cats had higher seroprevalences of Bartonella henselae and Toxoplasma gondii, probably due to greater exposure of feral cats to the vectors or hosts of these organisms.

Semi-owned or un-owned cats are less likely to have antibodies against coronavirus, the agent of feline infectious peritonitis (FIP), than are owned cats. Because coronavirus is transmitted primarily via the faecal-oral route, roaming cats' behaviour of burying their faeces may reduce the risk of transmission, compared with indoor cats sharing a litter box in a multi-cat household. Also semi-owned or un-owned cats usually live at a lower density compared with owned cats.

Most studies report a similar prevalence of infection with FeLV and FIV in semi-owned, un-owned and owned cats (see Luria et al 2004, and TNR programmes section E).

Nuisance complaints

Complaints about cats are often about their behaviours such as spraying urine, yowling and fighting (often at night), and fouling areas with urine and faeces leading to contamination of the environment. In addition, people do not like finding sick, injured, or dead cats, or remains of any prey items. Some of these objectionable behaviours are reduced or absent once the cat is sterilised.

Predation of wildlife

Wildlife predation is relevant because cats hunt small mammals and birds and the welfare of the wildlife involved is also a concern for the organisations that form the ICAM Coalition. Predation of wildlife is one of the most controversial issues regarding roaming cats (see Tantillo 2006, Slater 2005, Longcore, Rich and Sullivan 2009); the discussion here only introduces some of the main points of this controversy. Relationships between cat advocates and wildlife advocates can unfortunately be hostile with little dialogue between the two groups. Such polarisation is unfortunate as there may be some common objectives between the wildlife and cat groups, such as advocating sterilisation of cats, stabilising or reducing roaming cat populations, and finding ways of improving the environment for birds and mammals.

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Wild birds are common prey for cats

Data on predation by cats are limited, with studies involving relatively small sample sizes and focused on small areas. Nevertheless, some observations on cat predation can be made.

- On continents mammals are the main prey eaten by cats, with birds forming about 20 per cent of the diet (Fitzgerald and Turner 2000).
- Relatively few species of mammal commonly form most of the diet. While birds are a less frequent component of the diet, usually many more species are eaten (Slater 2005).
- In some conditions, such as on farms or in food storage areas, cats are valued as hunters of species such as rats, mice and rabbits.
- Cats living near refuse dumps or those that are fed by caregivers, households and others will have food scraps as a high proportion of their diet.
- Cats are opportunistic feeders, so providing them with a readily available food source as a part of a population management programme may reduce predation on their usual prey species; however motivations to hunt exist independent of end-goal consumption, so feeding is unlikely to stop predation completely.
- Wildlife that has evolved on islands with no mammalian predators is particularly susceptible to the impact of cat predation.
- The actions of humans have a much greater effect on vulnerable and threatened species than cat predation of wildlife.

There are also additional challenges with the current available research.

- Predation is often studied by examining the diet of cats in different locations. This is done by examining gastrointestinal samples from cats that are killed, faeces analysis, recording prey brought home by owned cats and examination of dead or partially eaten prey found in the environment. However, diet studies do not provide evidence of the impact on a species unless prey species abundance, potential repopulation rates, other sources of predation and mortality are also monitored, as well as the predation of cats on other predators of wildlife and the ecosystem in which the cats and their prey live.
- Sometimes findings from a study of a small unrepresentative number of cats at a particular time of year are extrapolated to the entire cat population of a country throughout the year, which gives an inaccurate representation of the magnitude of cat predation.
- Predation by cats of rabbits, rats, mice and other species considered abundant or problematic may be regarded as beneficial by some – the impact of desired predation on perceived 'pests' and valued wildlife is not always separated in studies.

Research on the hunting behaviour of cats before and after sterilisation would help to address some of the concerns about the impact of roaming cat predation on wildlife. Molecular genetic techniques that identify prey DNA in predator faeces are being developed; they offer an improved method for studying roaming cat food habits over time. The vulnerability of endangered species in isolated island settings where cats, as an introduced species are the main, and sometimes only predator, is of most concern. In this situation, co-operation and interaction between wildlife, conservation and cat groups is essential so that the optimal solutions for both populations can be found. These solutions may include a trap, neuter and return or relocate (TNR/R) programme aimed at reducing cat numbers (see page 26), a programme where all cats are caught and killed, or a combination of programmes. Eradication of a cat population, even on an island, can be very difficult to achieve and can be a costly, lengthy and unpopular intervention.

Background information

See **www.mammal.org.uk** for information on carrying out surveys of small mammal populations.
See **www.invasiveanimals.com** on the control of roaming cats in Australia.
See Slater (2005) for a more detailed discussion on the effects of feral cat predation on wildlife.

C. Components of a comprehensive cat population management programme: selecting the solutions most appropriate to your situation

An effective cat population management programme needs a comprehensive approach. Each programme should be tailored for a particular site according to the local cat population itself, local circumstances relating to cat ownership and wildlife sensitivity, and community wishes for the future cat population and their expected level of involvement in any programme. The data from the assessment phase described in section A should be analysed and interpreted with consideration of the influential factors described in section B to establish the local situation. The following section C outlines a range of components that might form part of a tailored and comprehensive cat population control programme.

Programmes must be planned for the long term. Companion animal population management is a permanent challenge because dogs and cats are valued by humans and hence a core population will be renewed and protected by people, providing a permanent source of potentially unwanted animals unless carefully managed.

Ideally, the overall programme should be coordinated by the local authority responsible for cat population management. NGOs should work with the authority to identify the areas in which they can support the programme and make most difference. It is possible, however, that there is little involvement of a local authority in implementation and the NGO will be working relatively independently on a day-to-day basis, but with authority approval.

Education

The first step in addressing the control of roaming cat populations is the education of the public (both cat-owning and non-cat-owning) on responsible ownership. The following factors should be considered:

- Public education to increase awareness of roaming cat issues and an impact on cat welfare will be necessary for engaging community involvement.
- Management strategies should focus on supporting responsible cat ownership and on reducing the un-owned and semi-owned population, with the aim of minimising the transition of cats from owned to semi-owned or un-owned and maximising the transition of cats from un-owned to semi-owned or owned.
- Education initiatives should be developed in coordination with the local education authorities and carried out by trained professionals. Development of key messages is an important first step and may be best achieved through multi-stakeholder consultations. These messages should be tested for their effectiveness and reviewed on a regular basis.
- Educational messages can be communicated in many ways, including through:
  - ➔ formal seminars and structured lessons in schools
  - ➔ leaflets and brochures provided to targeted audiences
  - ➔ awareness-raising in the general public through the press, billboards, radio and TV
  - ➔ directly engaging people in discussions and exercises as part of community-based programmes.
- All potential sources of education on cats should be engaged to ensure that messages are kept consistent. Ideally this should include animal welfare groups, the veterinary profession, schools, enforcement bodies and the media (including animal-focused media groups). It may be necessary for one particular body to take on a coordinating role.
- Veterinarians and veterinary students may also benefit from educational efforts, such as through learning about:
  - ➔ the rationale behind and justification for population management
  - ➔ their role in related public health issues
  - ➔ methods of reproductive control
  - ➔ key messages on responsible ownership for clients
  - ➔ euthanasia methods
  - ➔ how they can become involved with and benefit from proactive population management programmes that encourage the responsible care of cats, including regular veterinary care.
- It can take time for the impact of education on cat population management to become evident, so methods of monitoring and evaluation need to incorporate both short-term and long-term indicators.

Confining owned cats indoors

The decision to keep a cat indoors should be based on an assessment of the welfare of the cat whether it is kept indoors or allowed out, and the risks to animals that may be hunted by cats. A cat with access to the outdoors is at risk of traffic accidents, disease, trauma, theft or getting lost, whilst an indoor cat is at risk of not being able to perform some natural behaviours, and of social stress if kept with other cats in a restricted environment. Each location will have characteristics that impact on either side of this equation, and ultimately it is up to local authorities and the individual cat owner to decide the outcome. If owners decide to confine their cats indoors, it is essential

that they understand the requirements of their cats and ensure that their needs are adequately met; for example, the need to express certain natural behaviours and how this can practically be met indoors. This may include the provision of hiding and perching spaces and the opportunity to perform exploratory and predatory-type behaviours by hiding food and playing with toys. Hence the provision of reliable information on how to ensure good welfare of cats in an indoor environment is important (e.g. indoor cat initiative **www.vet.ohio-state.edu/indoorcat.htm, www.catalystcouncil.org** and **www.fabcats.org).** Owners who decide to allow their cats access to the outdoors are equally responsible for protecting their welfare through measures such as regular vaccination and parasite control, as well as considering partial confinement such as at night when the risks of traffic accidents are higher. Partial confinement may also be used to reduce the risks of predation by keeping cats indoors when prey is most active, such as at dawn or dusk, or at certain times of year when young animals are most vulnerable. Cat-proof fencing may also be used to restrict cats to gardens and prevent predation in the wider environment.

It is likely that confining cats indoors will not be a sustainable form of ownership in many countries where housing infrastructure is unlikely to be sufficient to confine a cat.

Legislation

It is essential that the cat population management programme fits within legislative guidelines – and is preferably well-supported by them. Legislation is important for the sustainability of the programme and can be used to ensure that cat population management is carried out humanely and consistently. Relevant legislation can be found at both central and local government level and is sometimes scattered within several different statutes, laws or acts. Separate policy documents may also be relevant and can impact on the emphasis or method of legislative enforcement.

Several issues need to be considered when using this component.

- There is a balance to be struck between clear legislation and legislation that is so restrictive and unrealistic that it does not allow for evolution in management practices over time.
- Time should be taken to draft new legislation carefully, drawing from the experiences of other countries and relevant professionals. An inclusive process with all relevant stakeholders participating should be used, including appraisal exercises where input is actively sought and incorporated from several sources.
- Changes to legislation are difficult to achieve so it is important that submitted drafts are accurate and realistic. The end product should deliver laws that: are holistic, sustainable and considered suitable and reasonable by the community; engage the authorities with their responsibilities; and succeed in achieving the desired impact for animal welfare.
- Sufficient time should be allowed for any changes to legislation to be introduced. Guidance notes should be provided in advance to help with interpretation.
- Legislation will be a ‘paper exercise’ unless it is enacted uniformly and enforced effectively. Effective enactment will usually require the majority of effort to be spent on education and incentives and the minority to be spent on carrying out punitive enforcement measures. Education about legislation has to be targeted at all levels, from law enforcement bodies (such as lawyers, police and animal welfare inspectors) to relevant professionals (such as veterinarians and shelter managers) and cat owners. Successful enforcement has been achieved in some countries through the use of animal welfare inspectors (also referred to as wardens or animal control officers). These officials are trained and resourced to provide education, handle animals when required and enforce legislation through advice, warnings, cautions and eventual prosecutions.

The term ‘ownership’ is harder to define from a legal standpoint. In welfare terms we can define both cat owners and caregivers by their obligation to provide for the cat’s needs. Under animal welfare legislation there is rarely any differentiation between owner and caregiver. When it comes to ‘duty of care’ both may be accountable.

A good example of well-measured differentiation is contained in the New Zealand Animal welfare (companion cats) code of welfare 2007 **(www.biosecurity.govt.nz/files/regs/animal-welfare/req/codes/companion-cats/companion-cats.pdf).** It makes a distinction between owner and caregiver and states that: “While a person who merely feeds cats in a colony is not the 'person in charge' in terms of the Act, and therefore is not legally responsible for the cats in the colony, it should be noted that, where people trap cats in the colony in order to provide for their vaccination, de-sexing or care, they will have legal obligations as the 'person in charge'."

This is further clarified in the accompanying report **(www.biosecurity.govt.nz/files/regs/animal-welfare/req/codes/companion-cats/companion-cats-report.pdf)**, which states: "The public draft of the Code sought to make people who provided care to cats in a colony legally responsible for the welfare of those cats. In terms of the Act, these individuals do not have the obligations of the 'owner or person in charge'. However, should they elect to trap the cats for the purposes of vaccination, de-sexing, etc., they acquire those obligations while the cats are in their care."

Registration and identification

Registration of cats is not normally required by law (some exceptions may apply, for example, in some parts of Australia and New Zealand). The most effective way of clearly connecting an owner with his or her animal is to use registration and identification together. This should encourage a sense of responsibility in the owner as the animal becomes identifiable as his/her own. Registration with identification is an important tool for reuniting lost animals with owners and can be a strong foundation for enforcement of legislation (including abandonment legislation and mandatory regular rabies vaccinations). Identification of owned cats should be promoted, but this is problematic compared to dogs, which are commonly identified by a collar and sometimes by microchip (see *Identification methods for dogs and cats: Guidance for WSPA staff and member societies* on **www.icam-coalition.org**).

It has been shown that collars are well-tolerated by cats and are a very effective method for visual identification (Lord et al 2010). Three types of collar were evaluated (plastic buckle, breakaway plastic buckle safety, and elastic stretch safety) and found to be equally effective. The collar should be carefully fitted, regularly checked, and the owner should be prepared to replace it if it comes off or is lost.

Sometimes cats are identified by a tattoo on the inner aspect of the ear (which is quite a small area). The cat has to be anaesthetised for tattooing to be performed, and the tattoo may not be easily read without closely handling the cat. Tattoos of the ear whose purpose is to identify the cat are different from tattoos whose purpose is to indicate that the cat has been sterilised (see page 25).

Identification by microchip ('identichip') is the preferred method for permanent identification, but it is relatively expensive, requires a certain degree of skill for insertion, is not visible and cannot be read from a distance as scanners are required to read the chip. Also, as with tattooing, there is a need for a registration centre with personnel to manage the scheme. Some owners may not notify the registry of a change in address so not all animals identified by a microchip or tattoo can be traced back to their owners. Microchipping has the advantage of being a global system, so animals moving from one area (or country) to another can continue to be identified. Before instituting a microchip system, it is advisable to check that the chips and scanners used conform to ISO standards. For example: **www.petlog.org.uk**

Mandatory registration and identification can help the practical problems faced by shelters. When a cat brought to a shelter is identified, it can be returned to its owner without delay. If not identified, it is by definition 'un-owned' so the shelter can implement its policies without the delay of waiting for an owner to come forward. Both scenarios will free up valuable shelter space, which will potentially increase capacity.

Identifying individual cats can be difficult because the majority of cats are non-pedigree and, at least compared with dogs, there is much less variation in size, coat colour and pattern and other physical aspects. It is often difficult to distinguish one cat from another i.e. a small black short-haired cat may look very much like another, so may not be recognised by its owner.

Control of reproduction

Control of reproduction is central to any population management plan. The following effects and considerations may need to be taken into account:

- Sterilisation reduces or eliminates behaviours associated with reproduction and also leads to better welfare of the individual cat (including being friendlier to humans, having a smaller home range in males, reducing roaming and fighting, reducing morbidity and mortality).
- It is beneficial to promote sterilisation of female and male cats before they reach puberty and are capable of reproducing; this also reduces the chance of mammary cancers in female cats in later life.
- Male (tom) cats should be sterilised as well as female cats because their sexual behaviour often leads to complaints from the public. Nuisance behaviours include urine spraying, howling at night, and fighting with other cats.
- Older males (more than 18 months to two years) have larger home ranges, and may be more successful at mating than younger males. All male cats should be included in a sterilisation programme.
- Voluntary, incentive-based measures may encourage owners to have their cats sterilised and identified (e.g. reducing the cost of sterilisation if the cat is microchipped at same time).
- It is necessary to ensure the availability of veterinary expertise to provide safe and effective sterilisation services.

Surgical sterilisation

Permanent sterilisation is currently achieved by surgery, where the reproductive organs of the cat are removed under anaesthesia (for guidelines see **www.sheltervet.org/index.cfm** and Looney et al 2008). Surgical sterilisation is relatively expensive and requires trained veterinarians and ancillary staff, an infrastructure (cages, building), drugs and equipment. If a local veterinary infrastructure already exists, it should be encouraged to offer a sterilisation service; financial investment as well as training may be necessary to ensure the service is taken up.

Non-surgical sterilisation

While currently it is most commonly achieved by surgical sterilisation, the development of effective methods for the non-surgical control of reproduction holds great promise for developing countries. While contraceptives are available for female cats, they are not licensed for long-term use and should only be administered under veterinary supervision. Long-term fertility control using chemical sterilants or contraceptives is an active area of research, but to date no such products are commercially available for cats. It is hopeful that they will be available in the future (see **www.foundanimals.org/index.php/About-Michelson/the-michelson-prize.html** for a discussion on funding available for such research and **www.acc-d.org** for an overview of available products and the research into chemical sterilants and contraceptives so far). Budke and Slater (2009) examine the use of matrix population models to assess a three-year single treatment non-surgical contraception programme versus surgical sterilisation in feral cat populations.

Vasectomy of male cats

As an alternative to castration in semi-owned and un-owned cats, it has been suggested that hormonally intact vasectomised males might provide better colony stabilisation. In observations by Stoskopf and Nutter (2004), however, vasectomised male cats showed no advantage over castrated males in stabilising colony populations. The time they stayed with a colony was similar to that of intact male cats, which was significantly shorter than castrated males. Vasectomised male cats had significantly larger home ranges than intact or castrated males and moved greater distances from the feeding sites. These findings are likely related to their search for breeding females, since the females in their home colonies were spayed. Vasectomised males also continue to perform behaviours that may be considered a nuisance, such as fighting and spraying.

Surgical sterilisation of owned cats

Because cats can be sexually mature from five to six months of age, and because unplanned litters in young cats are common, neutering just before puberty is commonly advocated.

While the usual age for neutering male and female cats is six to eight months, a policy of early age neutering (EAN) as of eight weeks of age (or when the kitten weighs at least 400 grams) should be considered, providing the necessary veterinary expertise is available and the kittens are in good health. Neutering at this age will prevent the accidental litters born to cats at the onset of puberty. In addition, owners are already in contact with the veterinary clinic for vaccination and parasite control and may be more receptive to having their kitten sterilised at that time rather than several months later.

In addition to identification, an owned cat should be marked to show that it has been sterilised. This is usually in the form of a tattoo on the ear in males and females (the preferred site) or at the surgical site in females. This mark indicates that the cat is, or was in the past, an owned cat, and is the equivalent of ear-tipping for un-owned and semi-owned cats managed by TNR (see section below). It also protects the cat from undergoing unnecessary anaesthesia and surgery again, for example, if it is caught as part of a TNR intervention or brought into a shelter by someone other than the owner.

Background information

See *IFAW Companion animal field manual* Part 4: Anaesthesia, Part 5: Technique for surgical sterilisation and Part 6: Surgical environment and post-surgical recovery. In the appendix there are protocols for EAN (part 6) and for assessing the need for post-operative analgesia (part 9). www.ifaw.org/Publications/Program_Publications/Companion_Animals/asset_upload_file726_61605.pdf

Detailed protocols can also be found in the Association of Shelter Veterinarians veterinary medical care guidelines for spay-neuter programs (Looney et al 2008).

Surgical sterilisation of semi-owned and un-owned cats

Sterilisation of these populations is usually delivered within a TNR intervention. Such interventions offer a non-lethal method of controlling semi-owned and un-owned cat populations, centred on sterilisation. Non-lethal methods are usually preferred by the public, and can be effective if interventions are well planned and implemented, and if the goal is stabilisation and eventual reduction in the size of the managed population rather than eradication.

A TNR or similar intervention should not be developed in isolation, but form an integral part of a comprehensive management programme that addresses all the issues identified as impacting on the cat population. For example, if the owned cat population is identified as a significant source of unwanted kittens, a TNR intervention alone will not efficiently impact on this source, a sterilisation intervention focused on owned cats, preferably including financial contributions from the owners themselves, should be included.

During implementation, there must be some flexibility so that the intervention can be adapted to deal with any problems or changes that arise in the cat population. For example, if a large proportion of trapped cats are found or suspected to be owned, an intervention directed at cat owners to sterilise their cats should be prioritised.

Because semi-owned cats have a higher reproductive success than un-owned cats, it is advisable to target this population before un-owned cats.

Trap, neuter, return (TNR)

TNR (sometimes referred to as CNR (capture, neuter, release) is a method of humanely trapping unneutered cats, neutering them, and returning them back to the same environment where they were collected.

Basic principles

TNR interventions will have a greater visible impact if focused on well-defined, preferably geographically restricted, cat populations, rather than diluting effort across multiple populations.

Managed colonies should not be maintained where cats are known to pose a threat to vulnerable species, near municipal water supplies, and other areas where they are very likely to come into conflict with humans or in regions where terrestrial wildlife rabies is epizootic unless widespread vaccination of cats against rabies is included.

The TNR location should give caretakers easy access and be safe for the colony (away from major roads) without attracting excessive attention from passers-by. TNR interventions can encourage the abandonment of owned cats at TNR locations, so a mechanism for identifying these cats and removing them from the colony for adoption should be in place as well as local education campaigns to discourage abandonment.

A TNR intervention should not be started if a significant proportion of the community is opposed, which could pose a threat to returned cats, or if there are no reliable caregivers to look after the cats. These factors should be addressed first, before including a TNR intervention within the overall management plan.

The concept of guardianship in TNR

Ideally, a TNR intervention should have a guardianship component where returned sterilised cats are regularly monitored and managed by a caregiver. Caregivers (also known as guardians or semi-owners) should provide a minimum level of care, which includes food and shelter, sterilisation, basic veterinary care (vaccination, parasite control), and veterinary attention in the case of injury or disease.

- TNR interventions can be aimed at both semi-owned and un-owned cats. By promoting guardianship, a well-run intervention should result in an increase in semi-owned and a decrease in un-owned cats. This may not be possible with all cats, as some will be too un-socialised or solitary-living rather than part of a colony.
- Cats should be monitored at least every other day, and preferably every day, by a dedicated caregiver who knows the cats and is committed to their welfare.
- The caregiver should trap and sterilise all new cats, remove tame cats (providing they can be re-homed) and trap and remove kittens young enough to be socialised (those less than eight to 10 weeks old) for adoption (providing they can be re-homed). In situations where re-homing is not an option, euthanasia of kittens should be considered if a high mortality, and hence suffering, is likely. If sufficient resources are available to limit mortality, these kittens could be left in place and trapped later for sterilisation, preferably early-age sterilisation if possible to avoid missing the first breeding cycle.
- A caregiver should have the permission of the landowner, or whoever is responsible for the location the cats are in, to manage the colony at that location. This can provide legitimacy and protect the cats and the caretaker.

The local community and authorities as well as NGOs and animal welfare groups can support the caregiver by providing:

- direct financial support
- free or subsidised cat food
- free or subsidised veterinary care, including sterilisation, vaccination and parasite control
- education on the best ways to manage the cats
- help with resolving any problems.

Excluding owned cats from TNR and similar interventions

In any cat management programme involving TNR, efforts must be made to exclude owned roaming cats from being caught in traps intended for semi-owned and un-owned cats. Owned cats must either be confined indoors during the trapping period, or be easily identifiable by means of a collar and tag, tattoo or microchip; they should also be subject to an additional intervention that aims to control their unwanted breeding. This can be achieved by alerting cat owners in the targeted area of the need to confine cats at certain times, provide collars for their cats, and/or acceptance that if their cat is trapped it may be sterilised. Methods to do this include leafleting, telephone contact, direct contact and local community meetings prior to the start of the intervention.

Nevertheless, some owned cats may inadvertently be trapped. If trapped cats are in good condition and friendly towards humans, this should alert personnel to the fact that they may be owned. Some cats benefit from a 12 to 24 hour ‘cooling-down period’; they may initially appear to be hostile and not socialised to humans, but will quickly settle down and become friendlier, and be recognised as owned.

The presence of a tattoo (in ear or at surgical site) indicates that the cat has been sterilised and is owned, or has been owned in the past.

A policy should be in place to determine what action to take with these cats. If they are not sterilised the decision may be to sterilise them, even without the consent of the owner (who should have been aware of the intervention and the need to confine or identify their cat). If they are already sterilised, options include releasing the cat where it was trapped (the assumption being it is owned and will find its way home) or taking it to a holding facility, where if it is not claimed it may be offered for adoption.

Types of TNR interventions

The most basic form of TNR involves free, low-cost or subsidised sterilisation (with ear-tipping) for roaming un-owned or semi-owned cats, as well as rabies vaccination (recommended for all locations where rabies is endemic or where there is a high risk of importation of rabies into a currently rabies-free area) and sometimes treatment against parasites.

Ear-tipping permanently identifies the cat as sterilised and protects the cat from the stress of being re-trapped and anaesthetised. In some interventions the left ear in female and the right ear in male cats are tipped; in other interventions the same ear in females and males is tipped or a notch is made in the lateral aspect of the ear. In general, careful tipping of 1cm from the tip of the ear is the preferred and best method of identifying a semi-owned or un-owned cat as having been sterilised.

TNR interventions can also include more focus on disease control and post-sterilisation care. For example, TTVARM: trap, test (for feline leukaemia virus (FeLV) and feline immunodeficiency virus (FIV)), vaccinate, alter (sterilise), return, and monitor (or manage). See section e for a more detailed discussion of testing for diseases and section b for a discussion of guardianship that forms the basis for monitoring.

In some cases, a cat colony has to be removed from a particular site, usually because it poses an unacceptable threat to a particular wildlife species or because the site is to be redeveloped. In this case, the R in TNR would stand for ‘relocation’ as opposed to ‘return’ and is an alternative to trapping and killing as a control method. However, it is not always a practical solution, especially for large populations due to limitations of space and cost, so it is only advised for small colonies in isolated situations.

It is important to ensure that whatever method is selected is consistently used and understood by the authorities, NGOs and the public, particularly if more than one body is involved in carrying out these activities.

Additional information on trap, neuter and relocate

- A new safe site, where the cats can be managed as a colony, must be found.
- Before relocating cats to a new site, it must be established that their presence there will be supported, is not contravening any regulations and does not pose a risk to other animals or to humans.
- All the cats must be sterilised, vaccinated against rabies (if appropriate, and other diseases as required) and treated for parasites after trapping and before being offered for adoption or relocation.
- All kittens or adult cats that are socialised to humans should be offered for adoption, with only un-adoptable animals transferred to the new site.
- The cats should initially be relocated to an enclosure or large cage and allowed to adapt to the new location and to the caregiver before being released.
- The colony is subsequently managed as a TNR colony.
- There must be a caregiver who is responsible for supervising the adaptation of the cats to the new site, and undertakes to provide adequate guardianship.

The orientation to the new location is crucial to get the cats to remain there. Cats should be confined in a large cage or similar enclosure for at least two to three weeks, until they consider that location to be their permanent feeding station.

Advice on relocation of cats should be sought from expert organisations (see www.hsus.org/pets/pets_related_news_and_events/san_nicolas_island_cats_042709.html and www.mrfrs.org/subpg/programmes/ferals.php and www.snip-international.org/ and www.alleycat.org).

Testing for FeLV and FIV

There is some debate about the need for FeLV and FIV testing, particularly if funds are scarce and such testing will mean that fewer cats will be sterilised as a result. In addition, the accuracy of positive tests for FeLV and FIV decreases when prevalence is low, so up to 50 per cent of positive test results might be expected to be false positives. Confirmatory testing is often impractical. The recent advent of FIV vaccination is an additional complication; the vaccine induces antibodies against FIV that cause false-positive results in the currently licensed FIV tests. While previously owned cats may have been vaccinated against FIV, it may not be possible to differentiate them from infected cats.

Most interventions choose to focus on mass sterilisation as the primary goal and do not routinely test for FeLV and FIV, because resources for managing cat populations are limited. Focusing resources on sterilisation will reduce the transmission of FIV (by reducing fighting) and FeLV (by reducing reproduction). One option is to test cats in an area for a period of time to establish the prevalence of FeLV and FIV, although this information may already be available from the local veterinary community; if prevalence is low, testing will not be worth the investment as the number of positives will be low as compared to the cost of testing and a high percentage of these positives will be false positives.

If cats are deemed suitable for adoption, testing for FeLV and FIV should be at the discretion of the veterinarian in charge. There may be situations where testing should be carried out, for example, in:

- socialised cats that are suitable for adoption but are showing signs of disease and a decision needs to be made as to whether to treat or not – if there is no option of treatment, cats should not be tested but euthanised
- socialised healthy cats that are suitable for adoption – if FIV positive they can still be re-homed but may require special care (e.g. being kept indoors); the lifespan of an FIV-infected cat can be as long as that of an uninfected cat.

Background information

Large epidemiological studies from the US and the UK indicate that FeLV and FIV are present in approximately four per cent of semi-owned and un-owned cats, which is not substantially different from the infection rate reported for owned cats (Levy and Crawford 2004). Male cats, especially semi-owned or un-owned, are more likely to be positive for FIV because it is transferred by fighting which often occurs during territorial disputes. FeLV requires close contact for transmission and is most commonly spread from mothers to their kittens. FIV infection has a different natural history than FeLV in that infected cats can often live for a normal lifespan.

Benefits of TNR

There are many benefits of TNR and similar interventions. They include:

Cat-centred

- Clumped food resources provided to managed cat colonies reduce or eliminate the territory defence associated with poorer, dispersed resources.
- Increased survival times of neutered cats relative to intact cats (Nutter 2005).
- Sterilised cats are more tolerant of other cats than intact cats so there is less fighting, in particular between males, leading to fewer injuries, reduced risk of disease transmission and hence improved health.
- Cats attempting immigration may be less likely to be excluded. This is a desirable rather than an undesirable effect, as the exclusion of cats may just lead to the establishment of colonies elsewhere which are not managed. When sterilised cats allow immigrant cats to join colonies, the new cats should also be subject to management.
- Sterilised cats, especially males, are likely to have smaller home ranges so are less exposed to risks such as road accidents.
- Improved health of females, who do not experience the risks of repeated cycles of pregnancy and lactation (Nutter 2005).
- Where kitten adoption is included in the intervention, this leads to increased longevity and survival of socialised and adopted kittens.
- Sterilised cats may become friendlier towards humans, which increases their adoptability.

Public health and nuisance-centred

- Fewer nuisance complaints due to reduction in urine spraying, mating, fighting and noise.
- Reduced complaints about sick or diseased cats and kittens in the community.
- Reduced risk of transmission of disease to humans or to owned cats (due to vaccination against rabies and other diseases, and parasite control).
- Cleaner environment (control of garbage and other potential food sources).
- Stabilisation and in some cases reduction in the size of the colony i.e. fewer cats, leading to less contamination of the environment with urine, faeces and food/prey remnants.

Wildlife-centred

- Stabilisation and in some cases reduction in the size of the colony i.e. fewer cats, leading to less predation.
- Smaller colony home ranges mean that although the high cat densities may result in the increased local impact of predation, the region affected is reduced compared to more widely-ranging intact cats.
- Intact female cats with nursing offspring are more prolific hunters due to the increased energy demands of nursing (Fitzgerald and Turner 2000, Nutter 2005). Spaying females eliminates reproduction and the associated increases in food requirements and hunting.
- Since breeding females and younger cats are more active and efficient hunters, the presence of sterilised aging cats may actually reduce predation (Nutter 2004a, 2005).

See the following resources and websites for extensive information on TNR interventions and related methods:

www.hsus.org
www.feralcat.com
www.alleycat.org
www.operationcatnip.org
www.forgottenfelines.com
www.fabcats.org
www.snip-international.org

Vaccination and parasite control

Preventative veterinary treatments should be provided to protect the health and welfare of cats and to reduce the incidence of zoonotic diseases. These treatments should be offered in conjunction with education about the other aspects of responsible ownership. Local veterinary communities should be consulted regarding the prevalence and distribution of infectious diseases and parasite infestations, so that a preventative treatment protocol can be tailored to a particular area and local circumstances.

Regular vaccination and parasite control is likely to improve the health of cats, and can lead to increased reproductive success. Therefore, a sterilisation intervention should be offered in conjunction with a preventative treatment service. As with sterilisation and contraception, preventative treatments can be used to encourage owners to accept the value of general veterinary treatment and population management tools. Wherever possible, the local veterinary infrastructure should be involved in providing preventative treatments, to ensure ease of access and continuity of treatment in the long term.

Vaccination

Vaccination guidelines are provided by the manufacturers of the product and are also available at several websites (see the American Association of Feline Practitioners (AAFP) guidelines, Richards et al 2006, **www.catvets.com/professionals/guidelines/publications/?Id=176 and www.wsava.org/VGG1.htm).** Guidelines may differ slightly from one another, and there is currently debate on which vaccines should be used, and the frequency with which boosters should be given.

Owned cats

In any location where expert opinion advises rabies control is necessary or desirable, healthy cats should be vaccinated at least once subcutaneously with a high quality inactivated rabies vaccine, as of eight to 12 weeks of age depending on the product label (ideally, vaccinating younger kittens is also advised if they are presented for vaccination and a repeat visit at eight to 12 weeks is unlikely to occur, as in the case of mass vaccination campaigns). A booster at one year, followed by boosters every three years or as required by local ordinance and the vaccine manufacturer, is indicated.

Wherever indicated by local disease prevalence, and based on a cost-benefit analysis, healthy cats can be vaccinated with a live attenuated vaccine against feline herpes virus, feline calici virus and feline parvoviral enteritis (panleucopaenia). Cats over 16 weeks of age are vaccinated twice subcutaneously three to four weeks apart, and cats less than 16 weeks of age are vaccinated as of six weeks, every three to four weeks until 16 weeks of age. Booster vaccinations should be given one year later, and then at three-year intervals. Pregnant cats and cats infected with FeLV or FIV should only receive killed virus vaccines.

Semi-owned and un-owned cats

Wherever possible, and certainly in any jurisdiction where rabies is enzootic or where vaccination for rabies is required by law, cats should be vaccinated against rabies following the principles for vaccination of owned cats. Cats managed by TNR or similar interventions do not usually receive more than one rabies vaccination, due to the logistical problems of re-trapping at a later date, but this may be sufficient to provide immunity for their lifespan, especially if a vaccine providing several years of immunity is used. Vaccination of colonies can result in a herd immunity effect, which is the point at which the proportion of immune individuals in the group is so high that the disease agent cannot spread through the population.

Whether cats are vaccinated for diseases other than rabies will depend on the intervention and local conditions.

The American Association of Feline Practitioners (AAFP, Richards et al 2006) advises the following for cats in TNR programmes:

- Rabies virus vaccines labelled for a three-year duration of immunity should be administered to all feral cats undergoing sterilisation in areas endemic for rabies.
- Vaccination of all feral cats against feline herpes virus, feline calici virus and feline parvoviral enteritis with live attenuated vaccines at the time of sterilisation is also highly recommended.
- An attempt should be made to re-trap cats for administration of booster rabies virus vaccines at one year and every three years thereafter. Booster vaccines for feline herpes virus, feline calici virus and feline parvoviral enteritis may also be administered at that time, but the need to boost these antigens in adult free-roaming cats is less clear.

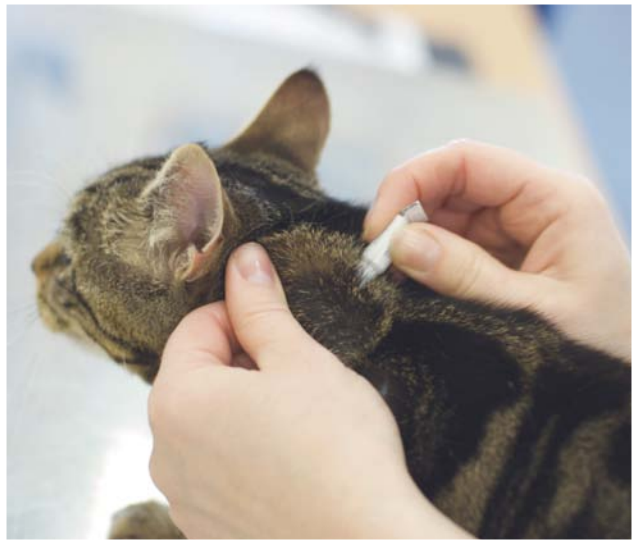
Parasite control

Parasitism is the most common transmissible problem of owned, semi-owned and un-owned cats. Common parasites include fleas, ear mites, ticks, intestinal ascarids (roundworm such as *Toxocara cati*), cestodes (tapeworms such as *Dipylidium caninum* and *Taenia taeniaeformis*) and hookworms (*Ancylostoma* and *Uncinaria* spp).

The choice of anti-parasite product will depend on the parasites to be treated, the route of administration that is possible, the availability and cost of the product and the characteristics of the local population. Wherever possible cats should be weighed accurately before dosage. Some products will not be suitable for pregnant and lactating animals. A specific protocol should be established for the treatment of kittens.

Agents that are given topically or by injection are most suitable for cats that cannot be handled easily, or those under anaesthesia or sedation.

See *IFAW Companion animal field manual* Part 7 Endemic disease and prophylaxis and Appendix part 7 Vaccination schedule and part 8 Anti-parasiticals dose rates.



Holding facilities, re-homing centres, and foster homes

Holding facilities and re-homing centres should be managed to a high animal welfare standard, and be designed to meet the cats' needs while minimising the risks of disease (see Haughie 1998, *ASV guidelines*). Such centres can be very expensive and time-consuming to manage, so adequate funds and personnel must be available to ensure their success. Awareness of the full costs required for re-homing centres is extremely important as centres are hard to close at short notice. Both capital expenditure and running costs should be considered and it is recommended that both the capital outlay and running costs for at least one year should be raised before commitment to a centre is made.

Centres should play an education role on responsible cat ownership within a community, in order to counteract any possible encouragement of abandonment they may represent. Owners of unwanted or problematic cats should be encouraged to consult shelters for advice and help, and to bring their cats to them rather than abandoning them outdoors or adding them to colonies.

Policies should be written to cover important topics such as sterilisation, re-homing, capacity and criteria for euthanasia. These should take into account the welfare of individual animals, the cost implications, the aims and objectives of the facility/centre and the impact of the facility/centre on the long-term management of the cat population. Protocols should be designed for each stage of shelter activities, from quarantine on arrival, to daily routines such as cleaning, feeding and exercise, to record keeping and re-homing practices.

The design of the centre should take into account the welfare needs of the animals and also consider public access, physical characteristics, services (such as drainage and water sources), potential noise disturbance, planning permission and potential for future expansion.

A network of foster homes may be an alternative to shelters, especially for kittens or for old cats who are particularly vulnerable to infectious diseases and/or require more individual attention.

See: Guidelines for the design and management of animal shelters, RSPCA International, 2006 and the Asilomar accords: **www.asilomaraccords.org/definitions.html**

Adoption of cats

Adoption is the ideal outcome for socialised kittens and cats and should be promoted whenever feasible. The success of an adoption intervention depends on the availability of holding facilities such as shelters or foster homes, and on the community's attitude towards adopting cats; if there is no tradition of owners obtaining cats from these places, the number of cats held will increase, leading to over-crowding and poor welfare. If negative attitudes exist towards adopting cats, an education intervention on the advantages of adopting cats from holding centres, shelters and foster homes should be in place before starting any population management programme.

While placing tame cats and kittens into good homes is the ideal, in some situations this can be difficult, as can finding suitable foster homes in which to socialise the kittens (those less than eight weeks of age). If homes are not available, or if there is no shelter or the shelter is full, there is the option of managing these cats as semi-owned, if a suitable environment and caregiver can be found. Long-term sheltering of cats should not be considered a viable option as their welfare is likely to be compromised when shelters struggle to meet their physiological and behavioural needs.

Euthanasia of cats

As part of all interventions, including TNR and holding facilities/re-homing centres, euthanasia will be required for cats that are suffering from an incurable illness, injury or behavioural problem that prevent them being re-homed or returned, or for cats that are not coping with their environment and are suffering poor welfare as a result. In preparation for responding to these situations, every intervention should have an agreed and written euthanasia policy. Ultimately, a successful population management programme should create a situation where these are the only occasions when euthanasia is required and all healthy animals can be found a good home or environment to live as a roaming cat. In reality, however, most countries will not be able to achieve this situation immediately but will need to work towards it while acknowledging that some healthy animals will be euthanised because not enough homes exist that meet the requirements of responsible ownership. Euthanasia deals with the symptoms and not the causes of population problems and must not be relied upon as a sole response.

Euthanasia of roaming un-owned and semi-owned cats will be the most humane option if they are living somewhere where they cannot stay and they cannot be transferred to another location or re-homed.

Adult cats that are not socialised to humans should not be kept in shelters except in cases of emergency, and a policy of euthanasia of cats in shelters should be used if they cannot be returned to an environment as a managed colony.

Only humane methods of euthanasia should be used. Intravenous injection of sodium pentobarbitone is the advised method, or intra-peritoneal injection of sodium pentobarbitone is an acceptable method of euthanasia for a cat that cannot be handled for intravenous access. The cat must be restrained in a squeeze-cage, trap or similar device and preferably should be sedated first (with an injectable preparation that leads to deep sedation and analgesia) before being injected intra-peritoneally with sodium pentobarbitone.

Kittens, unless newborns, should also be sedated prior to intra-peritoneal injection of sodium pentobarbitone.

See *IFAW Companion animal field manual* Part 3 Euthanasia and Appendix part 4 Euthanasia criteria (for medical reasons, behavioural reasons and due to inadequate guardianship). See also AVMA guidelines on euthanasia (formerly report of the AVMA Panel on Euthanasia), Schaumburg, Ill: AVMA, 2007. Available at: **www.avma.org/issues/animal_welfare/euthanasia.pdf**

Control of cat populations using lethal methods

Lethal methods of population control aim to eradicate or significantly decrease cat populations through culling a target number or proportion of cats. These are ethically questionable and are not popular with the public because of concerns for cat welfare and killing of sentient animals for perceived limited human benefit, hence alternatives should be pursued in preference.

Methods of lethal control considered unacceptable because of the animal suffering they cause include trapping followed by shooting or poisoning, shooting or poisoning in situ, hunting with dogs and deliberate introduction of diseases. All lethal control techniques can affect non-target cats (such as roaming owned cats) and other non-target species. See the **www.icam-coalition.org** website for the WSPA publication Methods for the euthanasia of dogs and cats: comparison and recommendations for a more detailed discussion of acceptable and unacceptable methods of lethal control.

Lethal control methods, usually aimed at eradication, can be successful if repopulation of the target area by immigration cannot occur, as with isolated islands. However, even in island situations, great effort over a protracted time is required to accomplish eradication. The majority of successful cat eradications have been achieved on islands less than 5km^2 in size, with populations estimated at an average of 40 cats per site and using a combination of lethal methods.

Although lethal methods can cause rapid depopulation, they are rarely effective in the long term in mainland areas. As long as food is available cats will potentially establish themselves in the empty niche. Removing cats, even at very high levels every one or two years, may not lead to a long-term reduction in their numbers because of repopulation through breeding and immigration. The presence of human populations ensures that a proportion of cats from the owned cat population will be available to reoccupy colony sites. Also, with almost any control method or combination of methods, a few breeding cats are left and they repopulate the area.

Protection of valuable resources like threatened and endangered wildlife in mainland habitats can be accomplished by cat exclusion measures such as predator fences. Exclusion fencing is considered to be the most humane non-lethal cat control method for wildlife areas, but the cost of establishing and maintaining fences can be prohibitive. Their use tends to be limited to the management of highly valued threatened species that live in relatively small areas.

See *Threat abatement plan for predation by cats*, Department of the Environment, Water, Heritage and the Arts, 2008, Government of Australia **www.environment.gov.au/biodiversity/threatened/publications/tap/cats08.html** and Slater (2005) for a discussion of lethal methods of control of feral cats.

Controlling access to resources

Cats will be attracted to areas where there are resources such as food. To control this, especially in areas where cats are not tolerated, restriction of access to the resources should be considered. However, this should only be done with specific localised resources and carried out very carefully in conjunction with measures to reduce the roaming cat population or provide alternative food sources in more suitable areas, in order to prevent cats from starving. Cat cafés are one popular way of providing alternative feeding sites for cats to limit conflict with people in a humane way, see **www.wspa.org.uk/Images/catcafe_leaflet_tcm9-2733.pdf** for more details.

Cat Café

D. Designing the intervention: planning, agreeing targets and setting standards

Once the components for the programme have been selected to suit the local conditions the full programme plan can be designed and documented.

Aims, objectives and activities

The programme plan should include clear and agreed aims and objectives including targets for activities such as the percentage of the cat population that is targeted for sterilisation or the number of cat owners in a particular location that will need to be reached by an education programme. It is also important at this stage to describe indicators that could be used to assess progress at each stage of the programme. The indicators will be used to monitor and evaluate the success of the programme and it is important to consider them at the outset as baselines will be required.

If a number of organisations are involved in cat population management, it may be relevant to draw up agreements so each party is aware of the overarching aim and their role within the programme. These plans should also be communicated to the end users, such as cat owners and stakeholders that will be affected by the programme even if they are not responsible for the activities themselves (this may include certain authorities).

Planning for sustainability

Cat population management programmes require human resources, infrastructure and finances. A plan of how the programme will be sustained in the long run should be drawn up at the outset; humane cat population management has a beginning but no end, as it requires ongoing activity to maintain the cat population in the desired state. Including and building upon local capacity will support sustainability, as will the development of responsible ownership as individual cat owners begin to support population management activities. It is important to consider the following factors:

1. **Responsibility:** Ideally resource requirements will be built into the budget of the responsible authority. Government bodies are most likely to be able to achieve sustainability through government funding, but may not regard cat population management as a priority. NGOs considering taking on responsibility for aspects of cat population management should ensure that they will be fully supported and resourced, whether by the authorities or other sources, before undertaking such responsibilities. They should also consider carefully that their investment will need to be long term and this commitment may challenge their capacity to take on other work.

2. **Owner involvement:** An intervention designed to have an impact on owner responsibility could lead to the sustainability of elements of the project, as well as permanent positive behaviour change. For example, sterilisation interventions could become sustainable if owners are encouraged to pay for this service, while at the same time the veterinary profession is supported so that it can provide this at an accessible price.

3. **Fundraising:** The ability to fundraise locally will depend on several factors, including the culture of charitable giving and the status of cats in the local community. Local people, businesses, trusts and cat-related industries (pharmaceutical, pet food and pet insurance) may all be interested in supporting cat management programmes, either financially or through providing resources (such as food or medicines). International grant-making bodies may also provide funding for specific project costs, but are unlikely to support long-term running costs. Again, the sustainability of each of these sources of funds and/or resources must be considered.

4. **Human resources:** There may be people willing to provide support through unpaid human resources, sometimes termed in kind or pro bono donations. Several professions carry out pro bono work for the benefit of NGOs, such as marketing, accounting and management firms.

5. **Veterinary professionals:** The veterinary profession is an important human resource, not just for surgical and medical skills but also for vets' ability to influence owner behaviour. Qualified vets may be willing to provide some regular services for free or at a low cost. Student vets may also be willing to help out as part of their training and this can become a formal part of their course, although supervision will need to be provided. Volunteer vets and vet nurses from overseas may also be a valuable source of support, although there is the potential for them to be considered a threat by local vets if they are seen to be replacing their services and there may be local legislation that prevents employment of foreign vets. The sustainability of this resource is also difficult as travel costs may be high.

It may be preferable to utilise these volunteer vets to support the growth and skills of the local veterinary profession.

6 **Registration:** A registration system with a small fee for cat ownership could provide funding for other components of the wider programme. However, the requirement for such a payment is an uncommon practice and unlikely to be easily implemented. The licensing of semi-owned cats living in colonies could be problematic.

Setting standards for animal welfare

The aim of maintaining the best practicable level of animal welfare should be clearly stated by the programme's standards. To ensure agreement and understanding, the standards are best developed by a team of stakeholders. Decisions regarding the fate of individual animals should be made on the basis of both their individual long-term welfare and that of the local cat population. There should also be a procedure for regular monitoring to ensure these standards are being upheld, as well as regular reviews of the standards themselves.

The following are common areas of cat management programmes that may require the application of minimum standards:

- surgery, including aseptic techniques, anaesthetics and drug regimes (e.g. analgesia)
- handling and transporting of cats
- housing and husbandry of cats
- re-homing procedures
- euthanasia – when euthanasia should be used and how it should be carried out
- record keeping and regular analysis of data – although not directly affecting animal welfare, good recording keeping can help identify parts of the programme that may be compromising welfare, for example, an unusually high incidence of post-operative complications at certain times may indicate the need for refresher training for certain veterinary staff or a change in post-operative care.

Minimum standards covering some of the above topics can be found in the *IFAW Companion animal field manual,* and in Looney et al (2008).

E. Implementation, monitoring and evaluation: checking the programme is achieving its goals

Implementation

This should be straightforward if priorities have been chosen sensibly and the design stage carried out in detail. This stage may require a phased approach, using pilot areas, which are monitored carefully to ensure any problems are tackled before the full programme is launched. The initial stages should not be rushed into. There might be 'teething' problems, and frequent updates will be required between key stakeholders to monitor closely and improve progress in the early phases.

Monitoring and evaluation

Once the programme is underway it will be necessary to monitor progress and evaluate effectiveness regularly. This is necessary:

- to help improve performance, by highlighting both problems and the successful elements of interventions
- for accountability, to demonstrate to donors, supporters and people at the receiving end of the intervention that the programme is achieving its aims.

Monitoring is a continuous process that aims to check the programme is going to plan and allows for regular adjustments. Evaluation is a periodic assessment, usually carried out at particular milestones to check the programme is having the desired and stated impact. Evaluation should also be used as the basis for decisions regarding future investment and programme continuation. Both procedures involve the measurement of indicators selected at the design stage because they reflect important components of the programme at different stages.

Monitoring and evaluation should be an important part of a programme, but it should not be overly time-consuming or expensive. Choosing the right list of indicators, with regard to their ability to reflect the changes that need to be measured – and to be measured with a degree of accuracy without too much additional effort – will be key to the success of this stage. In order to choose these indicators it is essential to have a clear plan of what the programme is setting out to achieve and why, and how the intervention will accomplish this.

Ideally monitoring and evaluation will be approached in a participatory manner where all relevant stakeholders are consulted and involved in making recommendations. It is also important to remain open-minded and positive during this process, as things may change contrary to expectations. The exposure of problems or failures should be seen as opportunities to improve the programme, rather than mistakes requiring justification.

The concept of monitoring and evaluation is not complex, but there are many decisions to be made regarding what to measure, how this is to be done and how the results should be analysed and used. These issues and others are discussed in much more detail in other texts, for example: **www.intrac.org.**

Monitoring and evaluation of all programmes should be achieved by regular observation, good record keeping, population-monitoring techniques using standardised protocols and periodic analysis of data. Methods include:

- surveys of the size of the roaming cat population (e.g. household surveys, participatory appraisals, mark-resight)
- surveys of the local community regarding the outcome of the cat management programme
- recording the incidence of nuisance complaints and public health issues such as cases of rabies in cats
- assessment of intervention activities and the observations of caregivers
- assessing the welfare problems of roaming cats (including the health and stability of managed colonies, kitten and adult mortality, disease incidence).

Evaluation of activities in shelters

If population management programmes are effective, they should have an impact on shelter activities. Measures that should be monitored include:

- the number of cats entering shelters
- where these cats are coming from in order to target the programme effort to problematic locations
- the proportion of cats that have some form of identification
- the number of cats reclaimed, adopted or killed
- the health of cats including disease incidence
- the number of kittens admitted
- the success of socialisation and adoption interventions for kittens and other cats.

Monitoring of TNR interventions

To ensure that a TNR intervention is being conducted appropriately the following measures can be used to monitor the success of trapping protocols used in TNR:

- the overall trapping effort – the number of trap-nights (number of traps times number of nights) until at least 90 per cent of the cats in the colony were captured or until no more than one cat remains un-trapped
- the trapping efficiency – the percentage of cats captured per colony.

Since cats that are part of a TNR intervention are marked by ear-tipping, the size of the managed population can be evaluated by a mark-resight study, provided that accurate records have been kept so the total number of cats treated, marked, released and predicted still to be alive can be estimated at the time of the resight survey.

Background information

Nutter et al (2004c) was able to achieve, for 107 feral cats in nine colonies, a mean overall trapping effort of 8.9 +/- 3.9 trap-nights per cat captured and a mean overall trapping efficiency of 98%+/- 4%. She attributed the success of the trapping intervention in part to the regular feeding schedules and locations maintained by the colony caretakers.

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The International Fund for Animal Welfare's community-led animal welfare manual is available at www.ifaw.org/ifaw_international/publications/program_publications/help_dogs_and_cats.php

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COMPANION & WORKING
ANIMALS UNIT

Identification methods for dogs and cats

Guidance for WSPA staff and member societies

Aim: Identification is a vital tool in the management of dog and cat populations. The aim of this document is to describe the key methods of identification suitable for dogs and cats, in order to aid decision-making and provide guidance on procedures and equipment. The advantages and disadvantages of each method and any inherent animal welfare considerations are discussed.

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Introduction

The world's population of domestic dogs and cats is estimated at one billion, with stray[1] animals thought to account for the majority. This considerable number of stray companion animals presents a serious concern for both human and animal welfare, and places a significant burden on communities. For any intervention that aims to manage and/or reduce this population, the importance of effective identification of dogs and cats should not be underestimated. Firstly, identification of owned animals significantly reduces the likelihood that they will become stray, especially when identification is coupled with registration as part of a successful registration or licensing system. Secondly, a suitable identification system is often an essential component of programmes that manage stray animal populations; enabling the recognition of animals that have passed through the programme (e.g. for neutering or vaccination) and the capture of useful data allowing the impact of the programme to be evaluated.

Identification of owned animals

There are many reasons why identification of owned animals should be encouraged, including:

- Identification is the key mechanism for reuniting 'lost' dogs and cats with their owners; hence it reduces the number of animals in shelters (and the financial and administrative burden this incurs to authorities or animal welfare groups), the number of healthy animals that are euthanased, and owner anxiety.
- Identification helps ensure that responsibility for an animal's behaviour can be correctly attributed, for example in the event of a dog attack.
- Visible identification discourages cruelty, theft and fraud, thus helping safeguard the welfare of the animal.
- By clearly connecting owner and pet, identification can reduce the likelihood of abandonment and encourage owners to take responsibility for their animal's behaviour.
- Identification can be an essential component of legislative systems that seek to ensure owners take adequate responsibility for their animals[2].

Identification is thus an essential tool in the effective management of the owned dog and cat population. This is especially true when it is coupled with registration: a system whereby details of individual animals and their owners are recorded on a central (usually national) database. Each animal is allocated a unique code, which is identified on the animal with permanent identification (a tattoo or microchip). When referenced to the database, the code yields information such as the owner's contact details. Databases can hold additional information such as the animal's vaccination status as part of a disease control scheme. In rabies-endemic countries, for example, registration and identification of owned companion animals is highly beneficial for the enforcement of mandatory rabies vaccination.

A registration fee can help cover administrative costs whilst also encouraging responsible ownership through financial incentives. For example, activities such as adopting from a rehoming centre, neutering, vaccinating, deworming and (approved) dog training may incur a discount or exemption. Any profit from registration systems can be rechanneled into population management measures and responsible ownership education.

In a number of countries identification and registration of dogs and cats is mandatory. This allows for the ownership status of animals to be determined immediately, reducing pressure on authorities and shelters. If owned, the animal can be reunited with the owner. It is important, however, that this is not seen by owners as a license to allow their animal to roam with a guarantee it will be returned. A reunification fee can discourage this as well as harsher penalties for owners who repeatedly lose or abandon their animals.

[1] In this document, the term 'stray' refers to dogs and cats that are on public property and not under the direct control or supervision of an owner.
[2] International Companion Animal Management (ICAM) Coalition (2007) *Humane Dog Population Management Guidance*. Available from www.icam-coalition.org.

Identification of stray animals (as part of an intervention)

Although identification can help distinguish responsibly owned animals, it should not be assumed that a dog or cat without identification is unowned. Any intervention in which dogs or cats are removed from the streets (whether or not to be returned) should consider that at least some of those animals will be owned. Every effort should be made to find the owner and engage them with the programme.

Visible identification methods such as ear notches or collars are widely used to distinguish animals that have been neutered, vaccinated or treated as part of a dog or cat population management programme. This can have the additional benefit of indicating to the community that these animals are being cared for as part of a management programme, raising awareness of the organisation or authority carrying out the intervention and potentially enhancing public cooperation.

Some methods may be less visible or not visible at all, but enable the identification of individual animals- namely tattoos and microchips. These methods enable much more useful data to be recorded during the course of an intervention, enhancing our understanding of the dynamics of stray dog and cat populations. Individual identification can also generate data for a comprehensive population survey[3], enabling more effective monitoring and evaluation. These methods do, however, need to be used in combination with visible methods of identification.

Choosing an identification method

There are several methods of animal identification appropriate for use with dogs and cats. Table 1 overleaf summarises the main methods used. In general, permanent methods are designed to last for the lifetime of the animal, semi-permanent for months to years (but are usually lost during the animal's lifetime) and temporary methods for no longer than a few weeks. Permanent methods can be coupled with registration on a database and offer enhanced security (against loss or falsification), and reliable certification (e.g. vaccination or legally permissible travel between countries). Non-permanent methods are usually cheaper, more visible and easier to administer, but less secure.

The identification method chosen should be the most humane and efficient of all available options, taking into account the target population, local conditions and the resources available. For population management programmes, the method chosen should be guaranteed to remain effective at least until the objectives of the programme are completed, and the animal should suffer no adverse effects on its immediate or long-term health or behaviour. The welfare implications of identification methods differ according to each situation, but the following factors should be assessed:

Capture, restraint and handling

- For nearly all methods of identification, some level of physical restraint will be required.
- Distress should be minimised by using techniques appropriate to the species, and bearing in mind the huge variety in size and shape of (dog) breeds.

The procedure

- The operator must be suitably trained (some methods require significant training).
- The time taken for the application should be minimised, but not at the expense of safety.
- Some methods can only be used on animals under general anaesthesia. Wherever possible, therefore, these methods should coincide with surgery (e.g. neutering), to minimise the time, cost and risk associated with performing general anaesthesia.
- Precautions must be taken to prevent the spread of infectious disease (either between animals or from animals to people).

Potential long-term implications

- Risk of infection or abecessation from skin damage or puncturing.
- Chafing from collars and injury caused by snagging or pulling of collars or tags.
- Toxicity e.g. from dye or ink.
- Ecological implications of increased visibility/altered appearance.
- Prolonged pain following procedure (e.g. due to growth restriction).

[3] For further information and guidance on conducting a dog population survey, see World Society for the Protection of Animals (2008) *Surveying roaming dog populations: guidance on methodology.* Available from http://groups.google.com/group/dog-population-survey-guidelines

	PERMANENT				SEMI-PERMANENT		TEMPORARY	
	Tattoo	Microchip	Ear Tip/Notch	Freeze Brand	ID Collar	Ear Tag	Paint/Dye	Radio Transmitter
RELIABILITY	* * *	* * * * *	* * * *	* * * * *	* *	* *	*	* *
COST	* * *	* * * * *	* *	* * *	* *	* *	*	* * * * *
VISIBILITY	*	–	* * *	* * * *	* * * * *	* * * * *	* * * *	–
LONGEVITY	* * * *	* * * * *	* * * * *	* * * *	* *	* *	*	* *
REQUIRES ANAESTHESIA	* * * *	* *	* * * * *	* * *	–	* * *	–	*
INVASIVENESS	* * *	* *	* * * * *	* * *	–	* * *	–	*
RISK OF HARM	* *	*	* * *	* *	* * * *	* * * * *	*	*
ACCURACY	* * * *	* * * * *	–	* *	* * * *	* *	*	* * * *
SPACE- RESTRICTED	* * * * *	–	–	* * * * *	* * * *	* *	* * *	–
UNIQUENESS	* * * *	* * * * *	–	* *	* * * *	* * *	*	* * * * *
INSTANT IDENTIFICATION	* *	–	* * * * *	* * * *	* * * *	* * * *	* * * *	*
TRAINING REQUIRED	* * * *	* * * * *	* * * * *	* * * * *	*	* * *	*	* * * * *
DATABASE REQUIRED	* * * *	* * * * *	–	* *	* *	* * *	–	* * * * *

* * * * * (Very high) * * * * (High) * * * (Medium) * * (Low) * (Very low) – (None / Not Applicable)

Table 1. Comparison of identification methods for dogs and cats.

Permanent Identification Methods

Tattoos

A tattoo is a mark created by puncturing an animal's skin and inserting a pigment (usually ink). This provokes an immune response, following which the pigment is left trapped within fibroblasts in the skin's upper dermis layer.

A tattoo is probably the oldest method of permanently identifying companion animals. It provides a permanent and effective way of individually marking an animal and hence is still widely used in dog population management programmes. However, as tattoos are not easily visible without close examination, they should be used in combination with another method such as ear notching (see Ear Tip/Notch), or collars (see Identification Collars) to allow visible recognition from a distance if catching is required.

Figure 1. Neutered and vaccinated dogs identified with ear tattoos as part of an Animal Birth Control programme in Jodhpur, India.

Figure 1 demonstrates how tattoos can be easily applied to dogs just after neutering, while they are still unconscious. On recapture, the tattoo can be referenced to records of where and when the dog was released, its approximate age, weight, condition and treatment. This can then generate estimates of survival and movement, and help monitor and evaluate the impact of the intervention. Some countries use a standardised tattoo symbol to indicate an animal has been neutered. In Australia, for example, the symbol Φ is tattooed on the ear of dogs and cats nationwide at the time of the neutering operation. Some organisations use a very simple form of tattoo marking to identify neutered females, by blotting ink over a freshly closed incision (so a permanent mark remains when the tissue scars over).

For owned animal identification, tattoos are largely being replaced by microchips, which are considered more reliable. The international Pet Travel Scheme, PETS[4], no longer accepts tattoos "*because tattoos can fade or become unreadable over time. Numbers on a tattoo could also be changed by further tattooing*." Furthermore, there is a lack of international and national control on tattoo identification to prevent duplication. Tattoos have traditionally taken the form of an owner's telephone number or social security code, causing obvious problems with changes in ownership. For this method to be successful in identifying owned animals, the tattoo itself should comprise a unique alphanumeric code, which is registered on a central database. The animal then wears a collar and tag bearing the registry contact details.

ADVANTAGES

- A tattoo is a secure, permanent form of identification.
- Tattoos can deter theft of owned animals. For instance, in the USA, the act of tattooing a dog changes its theft from a local misdemeanour to a federal felony (as the theft then falls under the 'Branded Animal Act').
- Unlike microchips, tattoos can be detected without specialised equipment.
- Individual animals can be identified with a unique alphanumeric code.
- Personal details of owners do not have to be on public display.
- Although the initial cost of purchasing an electric tattooing needle or a tattoo forceps kit may be high, the running costs of applying tattoos are low.

[4] The Pet Travel Scheme (PETS) controls the transport of pet animals across Europe and is legislated under the EU Regulation on the Movement of Pet Animals.

DISADVANTAGES

- Handling and restraint are necessary before tattoos can be located and deciphered (the degree of handling will depend on the location of the tattoo).
- Tattoos can fade and become illegible over time. This can have serious implications both for owned animal identification (e.g. reunification) and management programmes (e.g. data loss).
- Tattoos can be difficult to locate, especially on double-coated or long-haired animals.
- The procedure requires general anaesthesia (see Welfare Implications).
- Owned companion animal tattoos can often be arbitrary or illegible marks, with no useful function for identification.
- The procedure requires specialised equipment and a skilled, fully trained operator. The procedure can be time-consuming; needles have to be cleaned after each use, and (with tattoo forceps) the digits should be changed and tested before each animal is tattooed.
- Information is space restricted, e.g. by the size of a dog or cat's ear. If identifying a large number of animals, there may be insufficient space for the number of characters necessary to maintain a unique coding system.
- Tattoos can be removed or altered with additional marking or a laser, and ear tattoos can even provoke the removal or burning of the ear to remove the identification.
- This is not an immediate identification method (tattoos are generally illegible for several days after application).
- Legibility of tattoos is largely dependent on the equipment and skill of the operator.

WELFARE IMPLICATIONS

- The application of a tattoo is a painful procedure. WSPA recommends that tattoos are only applied whilst an animal is under general anaesthesia, for surgery, as controlled or supervised by a veterinarian.
- Tattooing requires puncturing of the skin, which can have infection implications and there is potential for disease transmission between animals. This risk can be minimised by ensuring all equipment and the application site is disinfected.
- The animal should be checked initially (and again on regaining consciousness) for signs of excessive bleeding, and ideally for the next few days for signs of local infection.
- The tattoo ink or paste used must be non-toxic.
- After a tattoo has been applied, a scab will develop at the site of application where the skin has been punctured, and there is likely to be initial redness of the skin. The site can take up to three weeks to fully heal.

PROCEDURE

The code

- Before implementing this identification method it is necessary to decide on a coding system. This should be devised on the basis of an estimate of the number of animals likely to be identified in the lifetime of the system or programme. **It is extremely important that each animal receives a unique tattoo.**
- Aim for maximum visibility of the marking within the available space. A combination of numbers (0 to 9) and letters (A to Z, excluding I, O and Q) yields the greatest number of permutations. Most tattooing forceps allow up to a six digit permutation of letters and numbers. One approach is to begin the code with a single letter followed by three numbers (e.g. A000, A001, A002 etc.) and after completing this sequence (i.e. after 999 codes have been issued) begin the code with the next letter in the alphabet. The letter could represent the year or month the animal was identified. This will allow for the identification of nearly 26,000 animals. Ensure a good supply of characters to maintain the order of your coding system.
- Each animal's code should be recorded in a database (manual or computerised), alongside a record of data. For owned animals, this will include details of ownership. For management programmes, this may include where and when captured, the date the animal was neutered, approximate age, breeding history, body condition score and any health problems.

The location

- It is important that the tattoo site is standardised when this is used as a system of identification. This will mean people know where to search when encountering an unknown animal, and in management programmes this will reduce handling time on recapture.
- The recommended location is on the **inside surface of the ear pinna** (see Figure 2). This is the most visible location, where a tattoo will be noticed even when not specifically searched for. The tattoo will not be covered by too much hair, and is less likely to be distorted by skin stretching during growth, making it more suitable for young animals.
- Another commonly used location is on the inside thigh of a rear leg. The stomach should be avoided as the tattoo may be later erased by surgery.

The method

The operator does not have to be a veterinarian, but must be adequately trained and competent. It is strongly recommended that tattoos are only administered on animals that are already under general anaesthesia (e.g. for neutering), which must be under veterinary supervision.

1. Remove as much hair as possible from the tattoo site. Remove any dirt, loose skin, grease or wax from the area and thoroughly clean using cotton wool soaked in surgical spirit or alcohol. Allow the area to dry.
2. Apply the tattoo using either a tattooing pen (see Method 1) or, for ear tattoos only, tattoo forceps (see Method 2). Ensure the equipment is sterile before use, and needles are suitably sharp so they will pierce the skin deeply enough to allow for the absorption of the ink. For ear tattoos, the ear should be held taut with no wrinkles.
3. **Do not remove excess ink from the tattoo site or attempt to clean the area –** this can reduce the effectiveness of the tattoo. Allow the site to dry and the ink will wear away naturally (this will take about 10 days).
4. After each tattoo, clean the equipment thoroughly to remove any blood, ink or other debris, and soak in disinfectant. This is important as needles are likely to become contaminated with blood thereby facilitating disease transmission. Dry the equipment thoroughly and store in a safe and dry location when not in use. Characters for tattoo forceps should be cleaned using a small, stiff brush (e.g. a toothbrush) and the forceps should be regularly oiled.
5. Record the animal's code on a record card and complete any other relevant documentation **after applying each tattoo –** avoid waiting and doing this in batches as errors may occur. For owned animal registration, tattoo operators need to provide the owner with a copy of the record card, and send another copy to the central registry within a specified time limit.

Method 1: Electric Tattooing Needle

This requires more skill than the next method (tattoo forceps), as the operator has to manually 'write' the inscription on the animal's skin, using a pen-style electric needle (see Figure 3). Hence, there is more scope for operator error in transposing the code, and there is also a lack of standardisation in the composition and appearance of the mark (e.g. how numbers or letters are written). With practice, however, this can be easier to apply and leave a more precise, fade-resistant tattoo.

Figure 3. Whilst still unconscious following a neutering operation, a dog's ear is tattooed using an electric tattooing device.

The device comprises an electro-vibrator system of needles that simultaneously pierce the skin and inject ink. The needle is either dipped in ink, has an automatic feed supplying ink to the needle, or there is a reservoir of ink in the tip of the needle.

An electricity supply is required for this method.

Method 2: Tattoo Forceps

Figure 4. Ritchey® ear tattoo forceps.

This method is the cheapest and easiest way of tattooing companion animals, with minimal training required for an operator to successfully leave a legible mark. This method is only suitable for ear tattoos, as access is required on both sides of the area where the tattoo is to be applied.

Specialised tattoo forceps consist of a pair of forceps or pliers, with individual metal tablets slotted into a frame in the jaws (see Figure 4). The tablets have protruding pins which, when the forceps are clamped around the ear, pierce the skin and leave a pattern of letters and/or numbers. Ink or paste is then rubbed into the punctures to produce a permanent mark.

The exact procedure will depend on the particular model of forceps used – instructions supplied by the manufacturer should be followed carefully, but these are general guidelines:

Figure 5. The ear of an anaesthetised dog is tattooed with forceps.

1. Open the lever lock bar and insert the desired characters into the forceps, taking care to insert them in the right order (in 'mirror' order). Close the lock bar.
2. It is advisable to test the forceps on a piece of card (the animal's record card for instance) to check the characters are inserted correctly and none are broken or too worn. This is important as it will be impossible to correct a mistake once the tattoo has been applied.
3. Shake the tattoo paste or ink to ensure an even concentration of pigment. Apply a small amount of liquid or paste ink directly to the tattoo site, using your thumb (or the roller dispenser of the container if applicable). Do not smear ink too liberally and obscure the veins. Apply some ink directly to the protruding pins of the forceps.
4. Before the ink on the tattoo site or pins has time to dry, position the forceps around the pinna avoiding hair, blood vessels or ribs where possible. The needles should be on the inside ear along the centre axis of the ear flap.
5. Pierce the ear through the ink with a quick, firm motion, avoiding excessive pressure but ensuring the forceps are fully closed, then release immediately (see Figure 5).
6. **Apply more ink or paste to the tattoo site (see Figure 6) and rub firmly (using thumb against fore finger) against the lay of hair into the puncture holes for at least 10 seconds to ensure penetration.** If there is bleeding, continue rubbing ink into the holes (if you use an ink with antiseptic or healing properties this will stem the bleeding). Do not remove excess ink.

Figure 6. Ink is applied to the ear of an anaesthetised cat that has been notched with tattoo forceps.

Fading and legibility

This seems to be influenced by a number of factors, including the location and proper preparation of the site, the procedure used, and the type and quality of ink or paste. If a tattoo mark is illegible it is likely it was not correctly administered. The following are some general rules for creating a longer lasting tattoo with tattoo forceps:

- Do not use turpentine or coal oil to prepare the tattoo site, as these will affect the ink.
- There should be three separate applications of ink or paste: to the site before application, to the pins of the forceps, and to the site after application.
- After the third application ensure the ink is thoroughly massaged into the puncture holes for 10 seconds.
- Tattoo forceps should be used properly with a firm, swift motion so that the punctures are of even penetration. If too much pressure is used, this can prompt profuse bleeding of the ear, which may continue after the ink has been applied and cause the ink to 'wash out' of the puncture holes.
- Ensure the forceps are closed completely when applying the tattoo – there will be a 'crunch' sound when this is done properly.
- Excess ink should not be removed, and if possible the area should remain dry and undisturbed until healed to prevent the ink from running (this can take several weeks).

Some other factors to consider are:

- A tattoo will not be clear for several days after it has been applied because of the presence of excess ink and the scab that forms where the skin has been punctured. It can take around three weeks before there is a clear, legible mark. Tattooing should thus be carried out some weeks in advance of identification being required and a temporary marker used initially.
- When a tattoo is applied, the pigment disperses down through the damaged epidermis and upper dermis layers. Its presence triggers an immune response, and phagocytes engulf the particles. As the area heals, the damaged epidermis flakes away, along with any surface pigment. As the upper dermis layer heals, pigment remains trapped within fibroblasts, concentrating in a layer just below the dermis–epidermis boundary. Its presence there is very secure, but over many years the pigment tends to migrate deeper into the dermis, and this can account for the long-term fading of tattoos.
- The uneven contours of the pinna, and irregular thickness especially near the base, may contribute to tattoo fading. Characters positioned towards the thin tip or fringe of the ear may be less defined, making the tattoo more difficult to read.
- The type and quality of ink used is critical in achieving a permanent, legible mark – see Equipment and Supply. The ink or paste should be highly contrasting with the colour of the animal's skin.
- Tattoos can fade in prolonged sun exposure. If this could be a problem then an alternative location such as the inner thigh may be more suitable (this will require the use of a tattooing pen).
- Forceps can hold characters of varying sizes. For dogs, characters around ¼ inch high seem to make the most readable ear tattoos.
- Tattoos remain the most legible when applied after the animal reaches adult height, and for ear tattoos there will be more room to correctly position the tattoo on adults. For puppies or kittens the inscription needs to be small and dark enough to withstand the distortion and fading caused when the skin stretches as the animal grows.
- In population management programmes, tattoos may be difficult to read in recaptured animals because of chronic skin infection or inflammation (which can cause darkening of the pinna) or scarring. The presence of ticks, other diseases or injuries can also obscure tattoos.

EQUIPMENT, COST AND SUPPLY

Tattoo needles/forceps

A tattoo forceps kit with characters and ink can be purchased for less than US$100. If looked after properly, the forceps will last indefinitely. Forceps specifically designed for smaller animals are available (see Figure 7).

Electric tattooing devices are more expensive, with prices starting at around US$150 (see Figure 8).

Tattooing ink or paste

- Tattoo ink or paste must be safe and non-toxic.
- The colour must be highly contrasting and is usually supplied in black or green.
- Ink and paste containing antiseptic properties should be used to reduce the risk of infection and stem bleeding.
- Ink sufficient for 50–60 applications costs around US$8, and a 500g tub of antiseptic tattoo paste costs around US$5.
- Ink may be supplied with a roll-on applicator (see Figure 9), which can be useful for application. This costs around US$3.
- 'Indian ink' (also known as India ink or Chinese ink) has found to be a long lasting option for animal tattoos. This is a simple black ink commonly used in printing or drawing. Verbal reports indicate that tattoos last 10 years or more with little or no fading, compared with 5 years or less for other types of ink. However, there is a concern that non-tattoo ink contains impurities or toxins which may lead to infection.

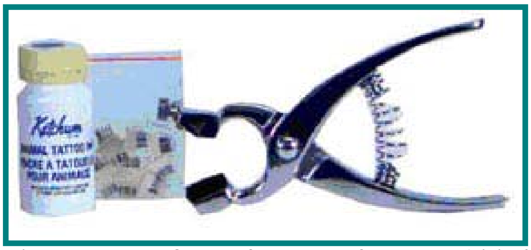

Figure 7. Ketchum® tattoo forceps, set of characters (digits 0 – 9 and A – Z), and tattoo paste.

Figure 8. Jorgenson® complete small animal electric tattooing kit.

Figure 9. Ketchum® roll-on tattoo ink.

Suppliers

- **Ketchum Manufacturing** (UK and Canada) guarantee tattoos will be legible and permanent if their equipment and ink are used and the correct procedure is followed. They supply a wide range of tattoo equipment (forceps, pins and ink). www.ketchums.co.uk www.ketchums.ca
- **Ritchey plc** (UK) and **Ritchey Tagg** (International). Formerly Brookwick Ward & Co.), this company based in Ripon, UK, specialise in animal identification and ship worldwide. They manufacture the Hauptner–Herberholz tattoo forceps and accessories (characters – 5mm, 6mm or 7mm in height, ink – in tubes of black, white or green, black liquid tattooing ink). They also produce special characters (5mm or 7mm). www.ritchey.co.uk http://www.ritcheytagg.com/ Ritchey plc, Fearby Road, Masham, Ripon, Yorkshire HG4 4ES Tel: +44 (0) 1765 689541.
- **Vet Tech Solutions** (UK) have a range of products for animal identification, including a complete electric animal tattoo kit. http://www.vet-tech.co.uk
- **Tattoo Supply Corporation** (USA) sells animal marking kits (Figure 8), which include the electric tattooing needle (with a tube and needle removal system for cleaning and sterilisation). They will ship internationally. www.tattooequipment.com
- **Animal Identification and Marking Systems** (USA) specialise in tattooing equipment for laboratory animals but sell electric tattooing pens suitable for dogs and cats. Useful training is also provided on their website. http://www.animalid.com/

The Microchip

Microchipping is a form of radio frequency identification (RFID), whereby a signal is transmitted between a transponder (the microchip) and a reading device (the scanner). The microchip is implanted under an animal's skin, and transmits a unique alphanumerical code when energised by the radio signal generated by the scanner. The scanner provides a digital display of the code, which is registered on a database along with information pertaining to that animal. The identification of companion animals is arguably one of the most useful and widespread application of microchips.

Figure 10. Identichip® microchip and accompanying tag.

The obvious disadvantage is that microchips do not provide visible identification; hence they need to be coupled with a visible method, preferably one that indicates the presence of a microchip (most microchip suppliers provide an accompanying tag to display on the animal's collar, as in Figure 10).

Microchipping does, however, provide the only truly permanent and effective method of identifying companion animals to date, with enormous potential for animal health and welfare. By providing such effective reunification this method significantly reduces the number of lost animals that contribute to the stray population. It also provides traceability to owners who persistently allow their animals to roam; encouraging responsible ownership. Microchip identification allows shelters to avoid the expense of housing, feeding, providing medical care, rehoming or euthanising thousand of animals, because of the reunification it facilitates. Microchipping is becoming a standard practice at rehoming centres, with many requiring all outplaced animals to be microchipped (i.e. before being rehomed or returned to owners). Microchips are the preferred identification method when considering the requirements of a standardised international system[16], and are now compulsory for cross-border pet travel under the Pet Travel Scheme (PETS) – no other methods are acceptable[4].

Setting up a microchipping system requires considerable planning and infrastructure, which must take into account the aims of the system and scope of animals to be identified. The high start-up costs can be prohibitive. Because of this, combined with the importance of central control and administration, it is strongly recommended that companion animal microchipping systems are administered by government agencies. This will also enable greater negotiation with microchip suppliers. Microchipping is often now incorporated under municipality licensing programmes, and in many countries microchipping is replacing tattoos as the government-recommended form of companion animal identification.

Incompatibility of microchips and scanners (a result of manufacturers developing competing technologies) has historically presented a serious limitation of this method. With the development of ISO standards this is becoming less of an issue (see Standardisation), and universal scanners are now available that can read most types of microchip. It is very important that only systems compliant with ISO standards are used – other systems may be cheaper, but in effect this devalues the whole principle behind microchip identification. When implementing this method consider the microchips and scanner as a package, to ensure compatibility.

Educating dog and cat owners about the importance of permanent identification with microchips can only be effective if those agencies dealing with stray dogs and cats have the equipment available to detect them. Otherwise, the rapid reunification offered by this method breaks down. It is important that organisations encountering dogs and cats not only have easy access to universal scanners but are aware of how to use them correctly and understand the importance of scanning every animal properly. In more developed countries, microchip scanners are becoming increasingly prevalent in rescue centres, shelters and veterinary clinics, and many animal control officers and animal welfare groups now routinely carry portable scanning devices. Microchip manufacturers often supply organisations with scanners free of charge, in an attempt to facilitate the use of microchips (and as an incentive to purchase that company's technology).

ADVANTAGES

- Provides permanent, unequivocal identification for the duration of an animal's life (a microchip typically lasts 25 years).
- Microchips cannot be removed or tampered with without surgical intervention.
- Microchips will not cause any discomfort if implanted correctly, and will not alter the animal's appearance or affect its behaviour.
- Minimal handling and restraint is necessary to identify a microchipped animal – physical contact is not always required.
- The implanting procedure is quick and should cause minimal discomfort (comparable to a routine vaccination).
- The technology has proven to be safe with reported complications rare.
- This is the only method that can guarantee a unique, unalterable code.
- Personal information of owners will not be visible to the public.
- Low margin of operator error (the code is pre-programmed and read electronically).
- Provides traceability which may infer wider benefits in relation to long-term monitoring and data collection, e.g. longitudinal studies of feral cats, alerting breeders to congenital defects (hence encouraging responsible breeding practices) and general companion animal statistics such as adoption and death rates.

DISADVANTAGES

- The technology required can be expensive to purchase.
- Supply may present a problem in some regions. Unless scanners are made freely and widely available to equip all agencies encountering stray dogs and cats, microchipped animals may go unidentified.
- A microchipping registry requires significant infrastructure, incorporating a computerised database and a 24 hour staffed call centre or website access.
- Identification is only possible if a suitable scanner is available.
- No visible identification. The microchip needs to be accompanied by a visible indicator or it will not deter theft of owned animals and may impede chances of reunification.
- Technology obsolescence remains a concern, i.e. not all scanners are universal and non-ISO microchips are still being manufactured.
- There is a risk of subcutaneous migration of the microchip from the original implantation site, especially in loose-skinned animals, although with more recent methods and technology it is unlikely that the chip will migrate more than a few centimetres.
- Comprehensive operator training is required (see Procedure).
- Risk of microchip failure, or of 'scrambling' if a second chip is implanted.
- The accuracy of this method is dependent upon the owner, e.g. in informing the database provider of changes in contact details or ownership.

WELFARE IMPLICATIONS

- If implantation is carried out properly the animal should feel no more discomfort than for a normal injection, and the whole process should take around 2 minutes.
- There is no requirement for either a general or local anaesthetic, or for hair removal.
- Any animal which appears to be in a poor physical condition or is known to react to needles or be allergic to vaccinations should be referred to a veterinarian before microchip implantation.
- There is a risk of introducing infection to an animal during implantation but this is minimised as the equipment is supplied in sterile units (see The Microchip).
- The scanning process may cause distress to some animals, especially as it is often necessary to scan the whole body of an animal, perhaps multiple times. This could be particularly problematic with fearful or aggressive animals. Certain animals may react aversively to the sound emitted by scanning devices (some scanners have the option to deactivate the sound).
- Once an animal has been microchipped and registered, there will be complete records tracking the animal's ownership status for the duration of its life, which will help protect the animal's welfare (e.g. when it becomes lost, or is involved in an accident).

- The safety of microchips has been reviewed by many regulatory authorities, as with any other implantable medical device, and microchips have been consistently approved as a safe and effective method of animal identification. Although there have been reports a link between microchip implants and tumour formation in dogs and laboratory animals, this has certainly not been proven. Referring to safety of microchips for human use, the US Food and Drug Administration stated that *"In all the safety data the FDA has reviewed for this device, including extensive animal data, we have seen no evidence suggesting toxic or carcinogenic effects."*[5] Although there is a need for continued scientific research into this technology, we know that many millions of dogs and cats have been microchipped worldwide with only a minute proportion reporting a problem – the World Small Animal Veterinary Association (WSAVA) Microchip Committee is aware of fewer than ten reports of tumours forming in association with a microchip[6]. WSPA believes that the benefits inferred by microchipping far outweigh this unsubstantiated risk to animal health.

THE MICROCHIP

Figure 11. Individual pre-sterilised microchip pack (PPS).

A microchip is an integrated circuit contained within a very small transponder, ranging in size from 12 to 28mm long and 2 to 3.5mm in diameter. The device is encased in biocompatible glass, so it can be inserted into living tissue without causing injury or adverse reaction, and can fit inside a compatible hypodermic syringe. The memory circuit of each microchip is programmed with a unique 15 digit alphanumeric code. The microchip itself is inactive with no internal power source – it is energised by the radio signal transmitted by the scanner.

Only microchips complying with ISO standards 11784 and 11785 should be used to identify companion animals (see Standardisation). Every microchip must be supplied in a 'pre-packed sterile' (PPS) unit (see Figure 11). The PPS unit should contain the microchip and needle, a sterilisation indicator, an expiry date and at least three self-adhesive bar codes containing the microchip's code (one each for the owner, the implanter's records, and the database). In some units, the microchip will be contained within a needle already attached to a disposable syringe (as in Figure 12). In others, the microchip and needle will have to be placed in a re-usable 'gun' (see Figure 16).

Figure 12. PPS unit including disposable syringe.

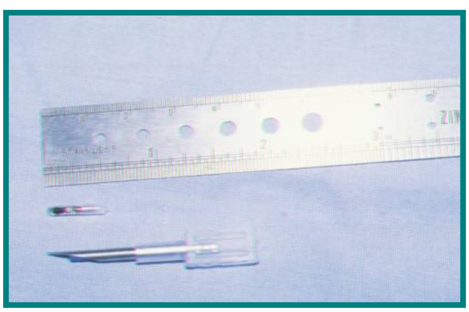

Figure 13. Microchip and implanting needle.

Microchips 12mm in length or less have been shown to be least prone to migration or breaking in independent trials[7]. The needle gauge and needle outer diameter are also important to consider. The needle needs to be larger than a conventional needle as it must house the microchip, but should be as thin and sharp as possible to minimise discomfort and the risk of haematoma upon insertion (see Figure 13). The supplier should offer to repurchase unused PPS units (to ensure their safe disposal).

Manufacturers of microchip technology should provide trace-back capability ensuring a specific microchip number can be traced from its source of production to the animal into which it has been implanted. Distributors should ensure they have full product liability insurance.

[5] Morrissey, S. (2007) Are microchip tags safe? *Time* www.time.com/time/health/article/0,8599,1672865,00
[6] World Small Animal Veterinary Association (WSAVA) Microchip Committee. Microchip Safety and Efficacy. www.wsava.org/Chip999
[7] Pet-Detect® Microchipping Equipment (2008) Technical Information- Migration. http://www.pet-detect.com/TIMicrochips.php

THE SCANNER

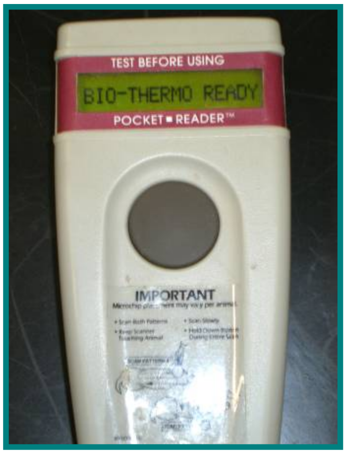

Figure 14. Portable microchip scanner.

Scanners generate a low frequency radio band that is intercepted by the microchip, which uses the energy from this band to power itself and transmit a return signal to the scanner. The scanner decodes the signal and displays the microchip's identification code on a liquid-crystal display window. With some scanners, the code can be relayed via computer interface to a database to instantly yield information about the microchipped animal (see Figure 14).

Scanners should have passed the ISO approved performance test (see Standardisation) and be able to read FDX-B (ISO 11784) microchips. They should also be compatible with the communication protocols in the geographic locale in which it is to be used, i.e. able to read microchips commonly used in the country. It is recommended that the scanner can read FDX-A microchips until 2026. Cheaper scanners will read fewer types of microchip. However, each additional technology to be read reduces the efficiency of the device's operation (e.g. speed, range etc), so the more expensive scanners should read most available microchips but may be slower to operate. Second-hand scanners should be avoided, as they may not be reliable.

Battery-powered scanners should have a low battery indicator and stop operating when operating function is compromised by low power input. Ensure batteries are always fully charged and that the manufacturer's directions for battery care are followed closely. As scanners emit and receive electromagnetic energy they may be affected by other electronic equipment or metallic objects and should be kept at least one metre away from these. If possible, avoid operating scanners on stainless steel tables and remove metal collars from animals prior to scanning.

PROCEDURE

Training

- In most countries microchip implantation is not considered a medical act and hence does not require a veterinary surgeon. Before setting up a microchipping system, however, it is important to check local and national regulations. According to the UK's Royal College of Veterinary Surgeons[8], the procedure does not need to be performed by a veterinary surgeon unless:
  – the method used is *not* the subcutaneous route, an ear tag or bolus;
  – the entry site needs to be repaired or closed;
  – the animal needs to be chemically restrained; or
  – there is any undue risk to the health or welfare of the animal.
  The American Veterinary Medical Association, however, categorises microchip implantation as a veterinary procedure, which must be performed by a veterinarian or under the direct supervision of a veterinarian[9].
- Whether or not they are a veterinarian, the implanter should be suitably trained by a competent organisation or professional. All companies following the ICAR Code of Practice (see Standardisation) provide training courses in the use of their products. Training should be comprehensive and include: the implantation procedure, potential adverse reactions, standard implantation sites, microchip technology, animal handling, health and safety issues and registration. A certificate should be issued to non-veterinarian agents along with an Implantation Manual, which confirms the individual attended and understood the course.

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[8] Royal College of Veterinary Surgeons (2007) Guide to Professional Conduct
[9] American Veterinary Medical Association (2006) The objectives and key elements needed for effective electronic identification of companion animals, birds and equids. http://www.avma.org/issues/pilcy/electronic_identification.asp

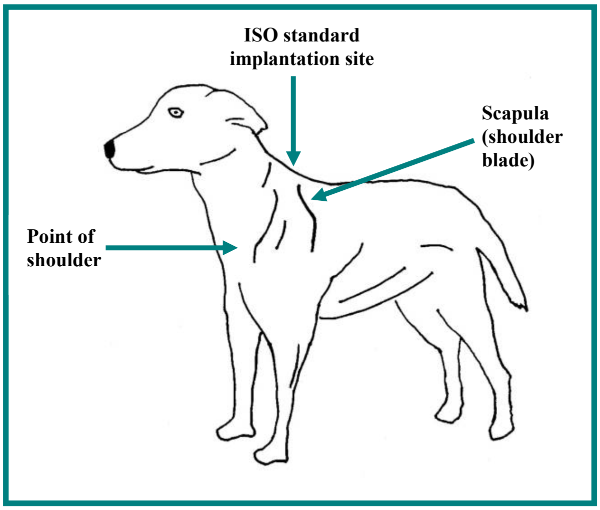

Figure 15. ISO standard microchip implantation site (at the back of the neck, midway between shoulder blades but slightly towards the head).

Implantation Site

It is crucial that the microchip implantation site is standardised for dogs and cats. For the signal to be transmitted efficiently between scanner and microchip the two devices must come into close proximity. This interaction will be optimised if operators know to target a standardised location, reducing restraint time and increasing efficiency.

The ISO standard implantation site for companion animals is **subcutaneously on the dorsal midline just cranial to the scapulae (shoulder blades)** (see Figure 15). This is also recognised by the WSAVA. The standard implantation site in continental Europe is in the midway region of the left neck, and this is the only other site recognised by the WSAVA.

Figure 16. Microchip implanting gun.

Implantation Procedure

Before implantation, ensure that the animal does not already have a microchip– scan the whole body slowly and carefully (see Scanning Procedure), and check for any other forms of identification such as tattoos. **If a microchip is already present do not implant another one**. A second microchip may confuse the scanner leading to mis- or no identification.

The microchip should be fitted according to the manufacturer's instructions (manufacturers should provide an Implantation Manual to each implanting agent), but the following are guidelines:

1. Ensure the packaging of the PPS unit is not damaged. Before opening the PPS unit, scan the microchip to confirm it is functioning and the code corresponds with the barcode on the packaging (this will also check the scanner is working).
2. The animal should be humanely restrained and comforted by a second person, as would be required for a vaccination. The second person should hold the animal's collar from the side while kneeling on the opposite side to the implanter, and facing in the same direction as the animal (Figure 17).
3. Clean the area around the implantation site thoroughly using a suitable antiseptic solution. This is essential for preventing bacteria from the surface being introduced to the animal during implantation.
4. Microchipping of dogs and cats is a simple straightforward subcutaneous (under the skin) injection. If the microchip is supplied already assembled with a syringe, simply remove from the PPS unit and it is ready for implanting.
5. For microchips not supplied in a pre-assembled syringe, open the PPS unit and remove the needle by taking hold of the plastic 'flag'. Insert the tapered conical end into the front of the microchip implanting gun, like the one shown in Figure 16. Once the needle is engaged, remove the plastic holder and retain for replacement over the needle after implantation.
6. Take hold of the skin just behind the implantation site. To ensure the microchip is left in the correct position, avoid pinching too much loose skin (Figure 17).

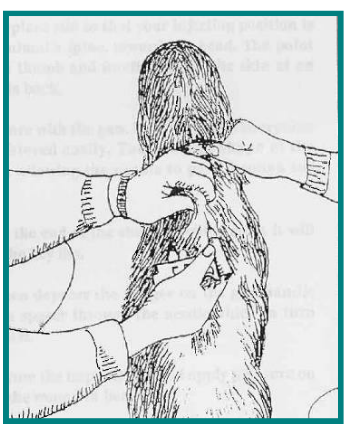

Figure 17. Implanting a microchip- one person restrains and comforts the animal, whilst another implants the microchip.

7. If the implantation site is between the shoulder blades your injecting position is with the needle parallel with the animal's spine, pointing towards the head. The point of entry of the needle should be between thumb and forefinger into the skin at a 30 degree angle. Insert the needle by applying gentle pressure. Entry should be achieved easily because the sharp, bevelled needle point will make a small cut in the outer layers of skin, allowing the rest of the needle to pass through. Push the needle in fully to the end of the shaft (to the plastic hilt if using a gun). If using a syringe, insert the needle fully and withdraw in a single smooth motion, whilst still pinching the surrounding skin. This and the angle of insertion minimises the possibility of the microchip accidentally being withdrawn with the implanting device. If using a gun, insert the needle fully and depress the trigger, then release. This will activate a plunger to push the spacer within the needle and expel the microchip into the subcutaneous layer of the animal.
8. Once the needle has been removed, apply pressure on the entry site with a finger for two or three seconds. The microchip should remain at a 30–45 degree angle to the longitudinal axis of the animal. (Over time, natural fibrocytes and collagen fibres will build up around the microchip anchoring it in location.)
9. Dispose of all needles and hypodermic syringes as clinical waste, in accordance with regulations.
10. Immediately after implantation, scan the implantation site to confirm successful placement of the microchip and ensure the device is functioning properly.
11. Complete the relevant documentation after implanting each microchip (see Registration).
12. Any adverse reactions or technology failures must be reported to the manufacturer and in some cases to a government body (e.g. in the UK the British Small Animal Veterinary Association (BSAVA) maintains a Microchip Adverse Reaction Reporting Scheme).

Scanning Procedure

Again, operators should familiarise themselves with the manufacturer's manual that should be supplied with each scanner, but the following are general guidelines:

1. Scanner function should be checked using a 'test' microchip, preferably an unused one, still in its PPS unit.
2. The scanner should be held horizontally, lightly touching the animal's fur, moving in small circular motions. Gently rocking the scanner from side to side will help locate microchips positioned at different angles.
3. Until a global standard implantation site is agreed on, scanning should concentrate on the site commonly used in that locality. If a microchip is not identified, a larger area should be scanned in slowly expanding circles for at least ten seconds.
4. The process should be repeated (with a different scanner if available) at least once before the animal is declared negative for microchip identification.

Figure 18. Scanning a dog for a microchip. It is important to scan the whole animal.

STANDARDISATION

When this identification method was first developed for use in companion animals, incompatibility of the various systems on the international market was a significant problem. Historically, manufacturers would only produce scanners capable of reading their own microchips. To overcome this problem, the International Organisation for Standardisation (ISO) developed two international standards, ISO 11784 and ISO 11785.

ISO 11784 is the standard that ensures the uniqueness of each microchip, by specifying the structure and information content of the 15 digit identification code it stores. Through a combination of country, manufacturer and identification codes, ISO 11784 offers countries or regulatory bodies a mechanism to ensure the uniqueness of international microchip identification. The first 3 digits of a 15 digit code that meets ISO 11784 will be either a country code or a manufacturer's code. The remaining 12 digits will be the identification code (ISO 11784 has reserved 274,877,906,944 possible combinations for the identification code). If a country code is used, it is the national responsibility of that country to ensure the uniqueness of these microchips. If there is no central database in operation, and microchips use a manufacturer's code, it is the responsibility of each manufacturer to ensure the uniqueness of those microchips.
The Country Code: The ISO has reserved 1024 possible combinations for country codes, which are defined under a separate ISO standard (ISO 3166: Codes for the Representation of Countries). Microchips that use a country code can only be used in countries that have one central database controlling the issue of identification numbers. This central database will allocate series of identification numbers to each manufacturer in that country, which can then produce microchips beginning with the three digit country code, followed by the 12 digit identification number. These microchips can only be sold within that country.
The Manufacturer's Code: In countries without a single central authority to control the issue of identification codes, a manufacturer's code is used. The ISO have appointed the International Committee for Animal Recording (ICAR) responsible for issuing the three digit manufacturer codes, and each manufacturer must apply to the ICAR to obtain one. In applying to ICAR the microchips will be thoroughly tested to ensure they comply with the ISO standards and the ICAR Code of Conduct, which includes an agreement on the part of the manufacturer to guarantee the uniqueness of the identification codes, and to allow full traceability of all animals identified. ICAR maintain a list of manufacturers who have signed the code of conduct on their website at www.icar.org.

ISO 11785 defines the technical aspects of communication between the microchip and the scanner. It permits either one way at a time signal transmission (Half Duplex) or simultaneous two-way signal transmission (Full Duplex). The new ISO regulation microchips operate at a frequency of 132.5 kHz, and are known as 'FDX-B' chips. When the ISO standards were introduced, there was an issue of 'backward' compatibility, in that the new scanners are still needed to read older, pre-ISO technology, for the duration of the lifetime of those animals. ISO 11785 was designed to address this issue in helping create a universal scanner that could read these new FDX-B microchips as well as the older technology such as FDX-A, HDX and FACAVA[10].

These standards were agreed by a number of international bodies including the World Small Animal Veterinary Association and the Federation of European Companion Animal Veterinary Associations, and were implemented in Europe in 1996. Since then there has been international agreement regarding the total transition to using ISO microchip technology[11], and this is now standard across Europe and Asia. South America and Africa are not yet a priority for most manufacturers in terms of companion animal identification, although this is changing[12]. Although manufacturers and distributors in the USA and Canada are able to implement ISO-standard microchipping systems in that region, unfortunately incompatible technologies continue to be produced and distributed, with the main impediment being how the cost and logistics of the transition to ISO-standard systems[12].

[10] Note: Annex A of ISO 11785 was developed to address this issue during the transition period between prior and ISO standard technology and defined the need for readers to read three technologies (Destron, Datamars, and Trovan) for a period of 2 years. AVID was not included in Annex A because they elected not to provide the encryption code with which to read their encrypted microchips. However, this 2 year period ended in 1998 and hence Annex A is no longer applicable, and Annex A microchips are not true ISO standard microchips.
[11] WSAVA (2002) Microchip Survey Results http://www.wsava.org/MicrochipSurvey1102.htm
[12] WSAVA Recommendations on Adopting and Implementing Microchip Technology that Adheres to the ISO Standards. http://www.wsava.org/MicrochipComm1.htm

The ISO standards and vigorous ICAR testing provide assurance that animals identified with an 'approved' microchip will be recognised by an 'approved' scanner. Under the Pet Travel Scheme (PETS)[4], if an animal's microchip does not meet ISO standards the owner must provide their own scanner when they travel. The standardisation effort has obvious positive implications for recovery of companion animals and disease control, and will lead to the more widespread use of this identification method. This, in turn, should hopefully help reduce the prohibitive costs of microchipping technology through the competition created by the standardised markets.

REGISTRATION

A microchipping system is only as good as its supporting database. After every microchip implantation, the microchip's unique code will permanently represent that animal and will correspond to a record on a central database where its details are stored. Without effective, reliable database administration, the system will fail.

When microchipping an animal the following information should be ascertained:

Essential details of animal:

- Microchip code
- Date of registration/implantation
- Species
- Breed
- Sex
- Colour/description
- Health-related information
- Spay/Neuter details.

If the owner is present:

- Full name and address
- Contact telephone numbers
- Name of animal
- Year of birth
- Special e.g. dietary needs
- Details of veterinarian
- Medical problems e.g. allergies, diabetes and any medication

The following requirements should be considered:

- Following microchip implantation, a record card must be completed (Figure 19), and the code and essential details need to be communicated to the central database, either electronically or by paper, as quickly and efficiently as possible. Ideally, all relevant persons (including veterinarians, other microchip implanters, municipal officers and animal owners) should be able to access the database and input new entries or update existing entries immediately as required, via a secure website.
- After implanting a microchip an accompanying visible indicator should be supplied. Microchip providers often supply a unique tag to attach to the animal's collar, e.g. Identichip (Figure 19). Another method is to tattoo an 'M' or 'X' on the inside ear (see previous chapter for tattoo procedure).

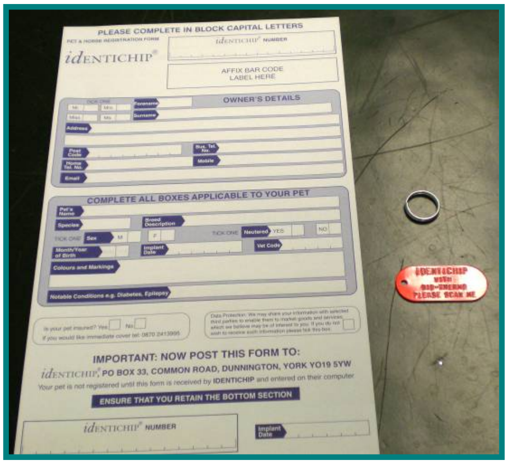

Figure 19. Identichip® registration form and tag.

- When registering owned animals a registration fee is usually charged, which covers the cost of the microchip and implantation, and registration. The fee usually falls within the range of US$30 to US$75. Often, the procedure is included in the adoption fee charged by animal shelters.
- The owner should be provided with a certificate of identification. In some countries, owners are enabled to change their own details on the database, which has proven successful.
- It is the owner's responsibility to contact the database service when information changes.
- The database should be fully accessible to all relevant authorities (including the veterinary profession, the police, municipality officials and NGOs involved in companion animal population management) 24 hours a day. Typically, database administrators provide a 24 hour, toll free telephone service, with the number advertised on the animal's tag.
- All records must be maintained at least for the life of the animal and be regularly backed-up. They should comply with national data protection legislation and codes of practice.

Many organisations are already operating substantial companion animal databases. These include ICAR member organisations and animal protection organisations, as well as governments and municipalities. According to ISO 11784, ideally every country should maintain databases of information about all issued codes and the associated animals.

Worldwide there are three main manufacturers or distributors of microchips used in animal identification: AVID; Schering-Plough (manufactured by Destron Fearing); and Trovan (the main European supplier). AVID is a large US manufacturer and its products are supported by a pet-tracking database called PETrac™. The other main database in the USA is the Companion Animal Recovery (CAR) database, which is operated by the American Kennel Club (AKC). Together these two databases identify and track approximately four million companion animals. Also, in the USA, Schering-Plough markets the HomeAgain™ Companion Animal Retrieval System. In the UK the register is kept on the Petlog database (jointly run by the Kennel Club, the RSPCA and the SSPCA), which can link with registers in other countries (useful for international travel).

Often in-country microchip administrators try to disseminate the message that there is one central point of contact, to speed up the process for all relevant parties, i.e. police, dog wardens and public. For example the UK's Petlog initiative, administered by the Kennel Club, administers a Central Microchip Reunification Service telephone number available 24 hours a day, seven days a week. In 2006, Petlog reunited 19,440 dogs and 11,285 cats with their owners, in some cases many years after the animal originally went missing.

Ultimately, what is required is a single international system that is technologically neutral. Having numerous proprietary databases (i.e. one or more per country) complicates the search process and could result in an animal not being identified. Europe is moving towards this idea and has established EuroPetNet – an online accessible database that records microchip codes from all European member databases. For data protection purposes EuroPetNet will provide details of the particular database in which a chip is recorded, but not the contact details of the owner.

Ear Tips and Notches

Ear tipping or ear notching is the removal of part of the distal pinna of one of an animal's ears, to provide visible identification.

Figure 20. Ear-tipped cat.

Ear tipping

Ear tipping (also referred to as ear 'clipping') is most commonly used to identify feral cats that have been neutered as part of a population management programme (see Figure 20). The procedure is carried out while the cat is under anaesthesia for the neutering operation.

WSPA regards ear tipping as the most humane, cost-effective and visible method of identification for feral cats in a managed colony. It is very important that all animal protection organisations working with feral cats use a universal identification method to signal that cats have been neutered. Identifying neutered cats (particularly females) can be very difficult without a visible indictor such as an ear tip, and unnecessary repeat surgery is both a waste of resources and traumatic for the cat.

This identification method should be used as part of a controlled programme in which cats are returned to a managed site where they will be cared for. Feral, unsocialised or ownerless stray cats that have undergone treatment as part of this programme should always be ear tipped before being returned to the original site or relocated to a new colony. For owned cats, owner consent must be obtained before ear tipping occurs. If a cat has definite signs of a previous neutering operation, it should be ear tipped, but if there is any doubt then the cat should be neutered and ear tipped. It is hoped that ear tipping will become a universal indicator of a neutered cat that is being cared for as part of a management programme. In the UK, for example, an ear tip is officially recognised in this way, and animal control officers know when they encounter an ear tipped cat that it is part of a managed colony.

Figure 21. Ear-notched cat.

Ear notching

This is similar to ear tipping but a small notch is removed from the side of the ear, rather than the tip, as in Figure 21. Ear notching of cats is not recommended as the mark may not be recognised by all organisations and may be mistaken for natural injury, e.g. from fighting.

Ear notching is, however, commonly used to identify dogs that have been neutered and vaccinated in a population management programme, often in addition to a tattoo. This identification method can be very useful in studying stray dog populations, if coupled with tattoos, as it provides an identifying mark visible at a distance. For example, when ear notched dogs are recaptured, either accidentally or deliberately, this can generate invaluable data on survival and movement, and other parameters essential for understanding the dynamics of stray dog populations.

ADVANTAGES

- Ear tipping creates a characteristic silhouette visible from a distance (this is particularly beneficial given the nature of feral cats).
- Ear notches can provide visible identification of dogs from a distance, depending on ear anatomy.
- The immediate visual identification provided by this method allows animal control officers and other agencies to recognise that a cat is part of a managed colony and is being cared for.
- Absence of an ear tip can immediately identify unneutered newcomers to feral cat colonies.
- Ear tips or notches can prevent animals being subjected to the stress of recapture, examination and unnecessary repeat surgery.
- Ear tips and notches are relatively easy to perform on animals already under anaesthesia e.g. for a neutering operation.

DISADVANTAGES

- In some countries, it may be illegal to carry out ear tipping or ear notching, or it may be subject to strict regulations. This is because the procedure may be categorised as surgical mutilation. It is important to check local and national legislation.
- The procedure can only be performed by a veterinarian, and the animal must be under general anaesthesia.
- Visibility may be reduced in long-haired breeds.
- The mark can be obscured by scar tissue or heal over.
- Identification is lost if the ear is injured or mutilated, and can resemble a bite wound, reducing reliability.

WELFARE IMPLICATIONS

- Ear tipping or notching is a form of mutilation and may cause pain or severe blood loss if not performed correctly. However, the procedure is not inhumane if performed by a qualified veterinarian, under sterile conditions, whilst the animal is already under anaesthesia for other surgery.
- The distribution of blood vessels in the ear means that there is a danger of excessive bleeding in dogs, hence the requirement for a veterinarian to perform this method.
- By providing permanent visual identification to indicate they are part of a management programme, ear tipping can be crucial for the health and safety of feral cats.
- This method can prevent the trauma of recapture and, potentially, unnecessary repeat surgery.

PROCEDURE

Ear tipping or notching should be performed by a veterinarian while the animal is under general anaesthesia (it is usually performed just after neutering). It is advisable to be consistent, as far as possible, with which ear is tipped or notched (i.e. left or right).

1. Clean both ear canals to remove any source of irritation which could provoke rubbing or scratching of the head.
2. Clean and disinfect the pinna then dry it completely to remove cleaning agents such as surgical spirit which could delay blood clotting.
3. Remove the tip or notch – general guidelines are provided in the boxes.
4. Apply digital pressure, a styptic or a drying antiseptic powder to the cut edge to halt bleeding – even flour is effective if there is nothing else available. If bleeding continues the cut can be sealed with tissue adhesive or electrocautery.
5. Check for bleeding when the animal recovers consciousness and blood pressure rises, and again before releasing the animal.

Ear Tipping

For ear tipping, the standard ear to be tipped is the left[13]. Approximately 6–10mm should be removed from the ear tip, depending on the size of the ear. If too much of the pinna is removed the ear appears 'cropped' and may be aesthetically unacceptable, but if too little is removed the mark will not be identifiable from a distance. The aim should be clear recognition at 20 metres.

Place a straight haemostat across the distal tip of the pinna, parallel to the base of the ear, exposing the area to be removed. Alternatively, mark a straight line on the inner surface of the pinna using a ball-point pen.

Cut along the line using sterile surgical scissors or scalpel blade (see Figure 22). Cautery can also be used although some report that this produces a ragged effect on healing. To create a distinctive silhouette it is important that the cut is as straight as possible and the tip looks artificial.

Figure 22. Whilst still unconscious following a neutering operation, the tip of a cat's left ear is removed.

[13] Feline Advisory Bureau (2006) Feral Cat Manual. *FAB Publication*

Ear Notching

Ear notches can be cut using ear notch pliers, which should be kept sharp and disinfected between animals. Alternatively, a thermocautery device can be used to cut a semicircular notch out of the leading edge of the pinna (Figure 23).

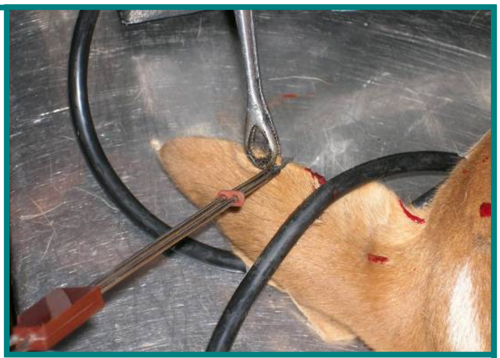

Figure 23. Ear notching of anaesthetised dog.

Freeze Branding

Freeze branding, also referred to as cryo-branding, is the application of a mark using an extremely cold branding iron. This method was originally developed for identification of livestock. When a cooled freeze branding iron is placed on the skin for the correct time and at the correct pressure, the extremely cold temperature destroys the colour follicles at the brand site, so they can no longer produce pigment, but the hair can continue to grow from the growth follicles. The result is that hair at the brand site contains no pigment and appears white, which can create a highly visible mark on dark-haired animals. If the iron is held against the skin for a longer period of time, then the growth follicles will also be a destroyed, and no hair will grow at all from the branding site. The latter technique is used for light-haired animals, as black or pink skin will be more visible than unpigmented hair.

Freeze branding requires the use of a coolant, usually liquid nitrogen although a mixture of dry ice and alcohol can used. This has obvious implications for human safety and the handling and use of such material will be subject to local and/or national legislation and regulations.

Freeze branding is widely used to identify horses, as it produces a highly visible mark. The effectiveness of freeze branding for use on companion animals is not widely known. In the USA apparently it is common for hunting dogs to be freeze branded in the ear or flank, and some report it is also effective on cats[14]. However there are a number of limiting factors relating to this method (outlined below) and WSPA would not recommend freeze branding for the identification for dogs and cats.

ADVANTAGES

- Highly visible on dark-haired animals.
- Mark is permanent – it will not fade or distort and is tamper-proof.
- A standard, uniform white mark produced (depending on skill of operator).

DISADVANTAGES

- Freeze branding is not a well recognised method of identification for dogs or cats, and is not acknowledged for legal ownership.
- Fine characters cannot be depicted – the mark is limited to the size and shape of the iron used, hence this method could not individually identify a large number of animals.
- The process is time-consuming. The iron can take around 30 minutes to cool to the correct temperature, and the branding procedure itself can take up to 10 minutes per animal.
- The procedure is complex, involving a number of different steps and equipment. The iron must be held against the skin for exactly the right amount of time – this will depend on many factors such as age, skin thickness, hair colour and the type of metal and coolant used. Operators must require training to master the technique. If the iron is in contact for too long, the animal will be scarred, but for light-haired animals the duration of the branding needs to be extended to destroy the hair follicles (the brand then takes the appearance of a hot brand).
- Liquid nitrogen and other coolant materials can be difficult to obtain.
- Freeze branding does not provide immediate identification – the mark will not be clearly visible for some weeks after branding.
- Freeze brands are not as visible on light-haired or white coloured animals, unless branding time is extended for longer, as described above.

[14] Farrell RK, Koger LM, Winward LDJ (1966) Freeze branding of cattle, dogs, and cats for identification. *Am. Vet. Med. Assoc.* 149(6):745-52

WELFARE IMPLICATIONS

- Freeze branding causes blistering of the animal's skin – the area will be swollen for a few days before a scab forms over the site. Whilst there is no puncturing of the skin, such blisters take time to heal (can be longer than a month) and can become infected should they burst.
- In a report of a working party set up by the UK's Royal College of Veterinary Surgeons to consider the mutilation of animals[15], freeze branding was found to cause 'minimal discomfort and no subsequent pain' in horses, but there has been no similar investigation for other companion animals. The procedure does, however, have the potential to cause pain, although some report that animals do not react to freeze branding because the extreme cold produces a numbing effect. According to the UK's Companion Animal Welfare Council, freeze marking does not require sedation or anaesthesia[16], but may cause pain '*depending on the skill of the operator, sensitivity of the animal and how long the 'brand' has to be held against the animal's skin*'.

HUMAN SAFETY IMPLICATIONS

- Great care must be taken when handling the coolant material, as this can cause injury because of the extremely cold temperature. Liquid nitrogen has a temperature of –240 degrees, and a dry ice – alcohol mixture will have a temperature of around –90 degrees. Precautions must be taken to avoid skin contact.
- Acetone and alcohol are extremely flammable and should only be used in open air or a well ventilated building, with no danger of open flames (e.g. smoking).
- Vapour from alcohol is dangerous to the tissues of the eyes and nose.
- The use of branding irons is subject to laws relative to each individual state or country.

[15] Royal College of Veterinary Surgeons (1987) Report of working part established by RCVS Council to consider the mutilation of animals. UK. www.cdb.org/vets/mutilations
[16] Companion Animal Welfare Council (2002) Report on the identification and registration of companion animals. UK. www.cawc.org.uk/sites/default/files/CAWCRepID&Registration02final.pdf

Semi-permanent identification methods

Identification Collars

Figure 24. Dog and cat wearing collars with identification tags.

A collar and tag is the most common identification method for owned dogs and cats. In many countries it is a legal requirement that companion animals wear collars displaying key information when in public. The owner's name and contact details, or those of a pet registry, and sometimes medical information, are inscribed either on a tag attached to the collar (as in Figure 24), or on the collar itself. Owners must ensure the information displayed by collars is accurate and up-to-date. Some pet registries will engrave a unique code on a tag to be issued when animals are registered, often to accompany a microchip.

Figure 25. 2008 'Red heart' rabies vaccination tag, provided by Ketchum Mdg. Co.®.

Collars can be an important component of compulsory registration schemes, as they are easily and cheaply administered, yet highly visible. For instance in countries where rabies vaccination is mandatory, collars or tags can be issued to owners at the time of vaccination (see Figure 25). The colour of the collars can be changed each year, providing a visible indicator that the dog or cat's vaccination status is up-to-date.

This identification method is, however, vulnerable to failure. Animals can slip through collars or they can easily be deliberately removed, and tags can become detached. It is therefore recommended that, for owned animals, collars are accompanied by permanent identification such as tattoos or microchips.

In dog population management programmes, a fabric or plastic collar provides one of the cheapest and easiest methods of identifying neutered and/or vaccinated dogs to prevent repeat catching. Childs *et al.* (1998), for example, found black plastic collars to be a useful marking method for use in distance sampling of rural dog populations[17]. Collars (or plastic tags attached to collars) can be colour-coded, for example to identify when the animal was captured, or in which locality. Collars can also be embroidered

[17] Childs JE, Robinson LE, Sadek R, Madden A, Miranda ME, Miranda NL. (1998) Density estimates of rural dog populations and an assessment of marking methods during a rabies vaccination campaign in the Philippines. *Prev. Vet Med*. 33: 207-18

or printed with individual codes. A note of caution, however, as collars can be removed or lost, which can bias statistics if not accounted for, e.g. a survival estimate will be lower than it should be.

Unlike ear notches, the public can recognise collars used in a management programme, especially if they are brightly coloured or embroidered, perhaps with the name of the organisation carrying out the programme. This can reassure the public that an animal is being cared for as part of humane, managed programme, or that it has been vaccinated against rabies. This can have the additional benefit of enhancing the visibility of the programme in the community, potentially improving public awareness and cooperation.

There are welfare concerns with this method, as animals can get caught by their collars, which may lead to strangulation or other injury. It is very important that collars used in management programmes are safely fitted and will not strangle the animal if it grows, gains weight, or becomes caught, i.e. it must 'give' and be designed to break or open without too much force. Collars should not be put on puppies or kittens. Another concern is collars enable people to grab hold of dogs, and may in this respect encourage or facilitate cruel treatment.

Collars are not practical for identifying feral cats, as they are often lost or need to be adjusted or removed. Cats try to remove or slip out of their collars and often get their front legs stuck part way through the collar if it is not fitted correctly. Re-trapping cats in order to replace, remove or adjust a collar is traumatic for the cats, and potentially dangerous for the caretakers who need to handle them. For owned cats, there is also the risk of removal or injury, but this is reduced because of owner monitoring, and the benefits of a properly fitted collar arguably outweigh the risk of injury.

ADVANTAGES

- Provides highly visible identification.
- No additional equipment or investigation necessary to identify owner, unless e.g. municipal rabies tags where owner details must be retrieved via the municipal authority veterinary clinic.
- Inexpensive.
- Readily available.
- Quick and easy to administer.
- Variable in type and colour.
- Instant recognition of dog/owner details.
- Can be colour-coded or inscribed to identify individual or groups of animals.

DISADVANTAGES

- Can be easily removed from animal, deliberately or accidentally.
- Can break or become caught.
- Collars and tags are subject to various forms of degradation e.g. rust or fading, and can become illegible.

WELFARE IMPLICATIONS

- Collars can become caught, which can result in serious injury and death.
- Collars allow an animal to be grabbed, which can be used both positively (e.g. recovering a stray animal) and negatively (e.g. by enabling cruel treatment).
- The size of the collar must be carefully selected in order to allow for normal growth and activity of the animal.
- Collars should be fitted tight enough to ensure they will not slip off, get caught, or result in irritating movement, but loose enough to not cause discomfort or restrict movement. As a general rule, leave enough space for two fingers to be slipped underneath.
- It is the owner's responsibility to regularly check the collar for fit and safety.
- Collars should be made from materials that are comfortable and safe, that do not absorb moisture and maintain flexibility in low temperatures.
- Collar width is important if a leash is to be attached. If too narrow, then excessive pressure will be applied to the neck, resulting in abrasion and pressure necrosis. If too wide, this may restrict movement.
- Modifications such as bells or light-emitting material may interfere with the normal behaviour and social interactions of the animal, although this may be deliberate, e.g. owners wanting to protect wildlife from cats.
- Certain types of collar are available which have inherent welfare concerns. Electric shock collars and anti-bark collars, for instance, are cruel and unnecessary devices and are illegal in some countries.
- Zip ties (plastic fastenings that tighten but do not loosen) must never be used as collars as there is a very high risk of strangulation.

TYPES OF COLLAR

Buckle / Flat / Quick release collars

- Standard identification collar, considered the safest type of collar if fitted correctly.
- Usually made of leather or nylon webbing, but other materials include polyester, cotton or hemp.
- Fastens with either a buckle (as on a belt) or a quick-release catch, and has an adjustable circumference.
- Identification tags or barrels can be attached, or details inscribed on the collar with embroidery or ink.
- Usually has a metal loop to which a leash can be attached.

Break-away collars

- These are similar to buckle collars but have an in-built safety mechanism allowing the dog to break free when excessive force is applied.
- Recommended for situations when a standard buckle collar could get caught and cause strangulation.

EQUIPMENT, COST AND SUPPLY

Figure 26. Hand engraving tool for metal identification tags.

Figure 27. VetScrive ™ automatic rabies tag engraver.

Figure 28. Identification 'barrel'.

- Many varieties of adjustable collars and tags may be purchased from pet stores, pet supply centres, feed stores, and other retailers that sell animal products.
- Collars should be made of materials which are durable; comfortable and safe for the animal; can withstand extreme environmental conditions; do not absorb moisture; and maintain their flexibility in low temperatures. Common materials used include flat nylon webbing.
- If identifying a large number of animals individually, plastic identification tags can be purchased, which are simple to assemble and durable.
- Alternatively, although more expensive, professional engraving hand tools or machines can be obtained to create rabies and ID tags. This enables instantly available identification details to be added to a collar, which can be created automatically (see Figures 26 and 27).
- Identification 'barrels' hang from the collar like tags, but the identification information is not visually apparent (see Figure 28). Instead, the information is recorded on a small piece of paper that fits inside the barrel. These may be preferable to collar identification or engraved tags, if the details are likely to change regularly, e.g. owners who move frequently.

Suppliers

- **Ketchum Mfg. Co.** (USA). Specialists in animal ID tags and rabies tags. http://www.ketchummfg.com/
- **NLS Animal Health** market the Vetscribe Rabies Tag Engraver (Figure 26), allowing the instant engraving of custom rabies or ID tags.
- **Non Permanent Pet Identification Products.** Useful site linking to a variety of identification product sellers including ID collars and tags. http://members.aol.com/TesterDesp/ID-other.html
- **Pet Tags Online** (USA). Supplies a variety of pet tags with fast international shipping. www.pet-tags-online.com
- **Mastergrave** (UK). Supplies pet tags and a range of engraving tools (Figure 26), starting at US$18. http://www.mastergrave.co.uk/catalogue/browse.php?product_Category_ID=117
- **Animal Instinct** (UK). Supplies the dog identification barrel (Figure 28) at US$3. http://www.animalinstinct.co.uk/acatalog/Pets_Products_Extending_Dog_Leads_12.html

Ear Tags

Ear tags come in a variety of materials and designs and are attached to the ear either through a purpose-made hole or via a metal clip. Tags can be used to identify individual animals with a unique code, or simply by the presence of a tag, which may be colour-coded. In some countries, proof of vaccination may be indicated by an ear tag.

Ear tagging is not a recommended method for the identification of companion animals because tags can be removed, fall off or become snagged, and there is a high risk of injury and infection.

Figure 29. Dog with metal ear tag.

ADVANTAGES

- Good visibility.
- Can identify individuals or groups of animals using a code or a colour.

DISADVANTAGES

- Tags can be removed accidentally or deliberately.
- Handling and restraint is required to read a tag number.
- Welfare implications due to risk of catching, injury and infection (see below).

WELFARE IMPLICATIONS

- Application of an ear tag that pierces the ear is a painful procedure and should be performed by a skilled and trained operator, whilst the animal is under general anaesthetic for another procedure (e.g. neutering), under the supervision of a veterinarian.
- Ears that have been tagged often become infected, especially if the tag is worn for prolonged periods and in hot climates, where it can cause fly strike.
- Ears with tags are more likely to be ripped or torn during fighting or grooming.
- Tags can be snagged e.g. on vegetation.
- When the tag is attached using an applicator that pierces the ear (rather than the tag itself) there is a risk of disease transmission if adequate precautions are not taken.
- Animals that have been ear tagged should be examined regularly to check for inflammation and other complications.
- Coloured ear 'streamers' and 'switches' are temporary markers sometimes used on larger mammals for greater visibility. They are usually attached through pierced holes or using metal clips. These can easily become caught on objects or be pulled by other animals, causing damage to the ear, and should not be used for dogs or cats.
- Tags that loop around the ear should not be used as they can easily become caught to objects, and do not allow for ear growth.

The RSPCA commissioned a study of the effects of ear tags on the welfare of sheep, which found that all ear tags result in an inflammatory response due to the wound created at insertion, but certain types of tags created less of a response than others. Metal tags caused major lesions, whereas flexible plastic tags caused less response[18].

WSPA does not recommend the use of ear tags for identifying dogs or cats. The welfare implications outlined above, particularly the high risk of infection, outweigh any potential benefits of this method, and there are safer methods available for the same purpose, such as collars.

[18] Edwards, D.S.; Johnston, A.M. (1999) *Welfare implications of sheep ear tags*, The Veterinary Record, May 29, 1999. Further research commissioned in 1999 by the RSPCA from A. M. Johnston, Royal Veterinary College, University of London.

Temporary identification methods

Paint or Dye

Paint or die can be applied to the fur of dogs and cats for temporary marking that will last from a few weeks to several months (depending on moulting rate, wear, and fade).

The method of application varies from simple painting to compressed air spray. Stencils can be used for identification of individual animals, or more remote methods can be used if this is not required (e.g. brush-tipped pole). The colour should contrast with the colour of the animal. The mark should be allowed to dry before the animal is released.

ADVANTAGES

- Can by a highly visible form of marking.
- Easy to apply with minimal or no handling and restraint required either for application or for identification.
- Both flanks can be marked (for ground observation) or the back (for aerial observation).
- Non-invasive.

DISADVANTAGES

- Not an accurate technique for identifying individual animals.
- Temporary, unreliable and cannot predict longevity.
- Not suitable for long-haired animals as fur will be matted by the paint or dye.

WELFARE IMPLICATIONS

- Only non-toxic materials should be used as paint may be ingested in grooming. Turpentine or tar-based paints can be harmful.
- Fur matting may cause skin irritation.
- May alter the animal's behaviour (e.g. increased time spent grooming) and the behaviour of other animals towards it.

EQUIPMENT, COST AND SUPPLY

- There are a range of non-permanent marking products available, depending on requirements (see Figure 30).
- Dyes commonly used are: Nyanzol A, Nyanzol D, blackpowder, clothing dyes, human hair dyes, red and orange aniline dyes, and picric acid.
- Should be non-toxic and waterproof.

Figure 30. Various paint/dye animal marking products, from left: spray dye, dye pressure sprayer (for concentrate), non-toxic paint stick, coloured foam marker, long-lasting animal marking sticks, high-visibility spray in fluorescent colours.

- **Raidex** (Germany). Manufacture a range of animal marking products. http://www.raidex.de/
- **Ketchum** (Canada). Suppliers of animal identification products including temporary inks. http://www.ketchum.ca/
- **Ritchey Tagg** (UK but ships worldwide). Specialises in animal identification products and stocks a range of marking materials. http://www.ritcheytagg.com/
- **Vet Tech Solutions** (UK) have a range of products for animal identification, including markers. http://www.vet-tech.co.uk

Radio Transmitters

The attachment of small external radio transmitters to free-roaming mammals has become a common method of monitoring the location and movement of individuals, and is used successfully in a variety of mammalian species, although is not widely used to monitor the dynamics of dog and cat populations.

This is a temporary method – the transmitter is designed to eventually detach from the animal or be removed. There are a number of ways transmitters can be attached to an animal, including adhesive, implantation, and collars.

This method requires expensive technology and should only be implemented when project funding guarantees the capacity to monitor the animals for the life-span of the transmitter and remove it when no longer required.

Transmitters vary in size, weight and predicted lifespan, and are available from several commercial outlets. The basic system includes a transmitter, power supply, antenna, material to protect the electronic components and a collar, harness or adhesive to attach the transmitter to the animal. Transmitter packages should be as light in weight as possible. The total weight (i.e. collar, transmitter, battery, aerial and bonding material) should be no more than 5% of the animals' bodyweight.

Pet Locators

Global Positioning methodology is now being used by dog and cat owners to keep track of their animals, in case they roam or become lost. A lightweight, waterproof locator device is attached to the collar, and alerts the owner (e.g. via text message) when the animal leaves the radius of a specified location. The owner can then identify the animal's location on a computerised map, or by calling a helpline. This system has the potential to reduce the number of owned dogs and cats that stray.

Although cost-prohibitive, the system has potential for use in studies monitoring the movement of stray dogs, allowing continuous tracking.

A GPS pet locator can be purchased for approximately US $200 (e.g. http://www.zoombak.com/products/pet/).

Future identification methods

Improvements in technology will allow other methods for the identification of dogs and cats in the future, as well improvements in methodology for some of the existing methods. Reliability, visibility, minimal invasiveness, accuracy and cost are the primary considerations.

DNA Profiling

DNA profiling is now well established as a reliable means of identification for dogs, and this may be the future for the identification of owned companion animals. An animal's DNA profile is unique and hence allows unequivocal individual identification. Also, because it is permanent, it can never be altered, modified or removed. Although the technology for this method is very complex, it is simple and non-invasive. Animals can be identified through samples of saliva, hair, blood or semen. If their DNA profile is then registered on a database, this will provide a permanent record via which the animal can always be identified. As with other permanent identification, it will be the owner's responsibility to ensure the database is up-to-date.

Currently DNA profiling is used primarily by dog breeders to provide proof of a dog's pedigree lineage, and also provides a genetic health screen for a number of genetic diseases and hence has the potential to improve the genetic health of dog populations. Knowledge of genetic disease transmission and carriage will enable better management of canine health.

Recently, scientists at the University of Cambridge in the UK, in collaboration with a genetics company Blueprint Healthcare Ltd, have developed new technology for canine DNA profiling that combines identity, pedigree and disease testing. Blueprint Healthcare Ltd can now offer DNA testing direct to dog owners and dog breeders. The DNA profiling service based on this technology was launched in the UK under the brand name DNAtag™. Blueprint Healthcare Ltd will be distributing swabs to all vets, police and animals shelters across the UK, in order to offer a free DNA profiling service, which will facilitate the speedy reunification of lost and stolen dogs. Presently, samples have to be sent to a laboratory for examination but when tests become portable they will presumably be more widely used.

RFID 'Invisible Ink'

In 2007 the US company Somark Innovations announced the development of a biocompatible 'RFID ink', which can be read through animal hairs. This passive technology could be used to identify companion animals using radio frequency identification but without the need for implanting microchips. The company is planning to licence the technology to markets that will include companion animal identification.

The ink in effect forms a fake fingerprint. It is apparently safe for human and animal use, contains no metals and is chemically inert. The ink can be either invisible or coloured, depending on requirements. The application process takes between 5 and 10 seconds and involves a geometric array of needles, an ink capsule, and a re-usable applicator. The ink is 'tattooed' on the animal with no need for hair removal. The ink remains in the dermal layer and can be read from four feet away. The amount of information the ink can contain depends on the amount of surface area available. In cattle, it is proposed that the ink will contain a 15 digit number, which can be linked to a database that contains additional data.

Retinal Scanning

Retinal scanning identifies animals by the distinctive vascular patterns at the back of the eye. This is generating interest as a potential identification method for animals because it is a non-intrusive identification process with almost 100% accuracy and declining cost. Studies indicate that changes to the retina over a dog's lifetime will not preclude positive identification of individual dogs by use of their retinal vascular patterns[19]. The technology is currently being developed by Optibrand but is still in the research phase.

[19] Gionfriddo, J.R. et al. (2006) Evaluation of retinal images for identifying individual dogs. *Am. J. Vet. Research* 67(12): 2042-2045

IFAW International Fund for Animal Welfare

FIELD MANUAL

MANUAL

Field Manual of Veterinary Standards for Dog & Cat Sterilization Surgery and Anesthesia

February 2014

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Foreword: Welfare in veterinary care

Appropriate and timely veterinary care is one of the fundamental welfare requirements for animals who are dependent on human guardianship. Within the provision of veterinary care, great attention must be given to minimizing the stress that an animal suffers. Stress over which an animal has no control is severely detrimental to the emotional well-being of the individual, and compromises all the processes that we endeavour to support with veterinary care: healing, recovery from illness, and physiological stability.

By nature of requiring veterinary treatment, animals are already suffering. They may be ill or in pain. They are often frightened and confused. Their ability to control the responses of their bodies and minds to environmental challenges may be compromised. They suffer metabolic and emotional stress to heal and to fight disease.

In the process of administering veterinary care, we must be aware of the many stressors that we may add to an animal who may already feel unwell. We subject animals to a great deal of human handling to which they may not be accustomed, or which they learned from previous experience to be punitive. When we catch animals for sterilization projects, they are frightened, confused and severely stressed, as anyone would feel on being trapped in a cage or a net and carried off. At vaccination clinics, dogs and cats may be hauled in by their owners by all means of force and transport, and can smell the terror of other animals all the way across the village. The more the animal screams and urinates and claws and snaps in complete and abject panic, the more brutally people who haven't learned better may respond. In a clinic, there are smells of blood, illness, stress and chemicals. There is a great deal of human activity. Lights may be bright, fluorescent, and on at unnatural times. There are sounds of machines, doors, human voices, distressed animals. Animals are confined to cages and forced to submit to our schedules and imperatives. All of these, particularly the latter, compromise one of the most critical elements of the welfare of any living being: the element of choice.

The feeling of choice is to be aware of options for how to respond to one's environment. Taking away someone's choices is one of the most profound stressors that we can imagine. Our societies use the restriction of choice by imprisonment to punish people. Caged, sick animals who must endure human handling on a human schedule have very little feeling of control. Many of the animals with whom we work in field projects come from free-roaming or previously abused lives in which confidence with humans played little role. The stress that these animals experience under clinical care, much less with uncompassionate handling, may be profound.

To provide veterinary care, we cannot avoid handling animals, or temporarily to cage them, or to give them drugs that may make them feel woozy, or to subject them to stressful procedures. But we can do a great deal to mitigate the stress that animals suffer while they are under our care. In every direct or indirect interaction with an animal, it is important to be empathetically and cognitively aware of the stressors that he or she may be suffering, to anticipate and pre-empt them, to relieve them as well as possible, and to de-escalate fear and anxiety. Even if animals are under veterinary care for only a day or two, or for only a few hours as in trap-neuter-release programs, or for a moment to vaccinate, minimization of stress and addressing welfare needs of patients is paramount to professional veterinary practice and to the recovery of animals from surgery or illness.

Protocols and standards described in IFAW veterinary documents often include points to address animal stress. However, these documents must not be taken to describe stress mitigation in exhaustive detail. Understanding stress and integrating stress management into every aspect of veterinary care is paramount to the practice of the profession, and is assumed to be recognized as such by practitioners. Demonstration of these concepts is also central to veterinary training.

Under no circumstances may we compromise welfare standards with the rationalization that animals are "stray" or "are used to a hard life". Animals are not lesser beings for lacking adequate human guardianship, nor do they suffer less. Animals must never be subjected to substandard welfare during veterinary procedures or for research, or to unnecessary risks for students to practice skills. It is our responsibility and privilege as veterinarians and as welfare guardians to ensure that all animals are held in equal respect.

Kati Loeffler, DVM, PhD
IFAW Veterinary Advisor
March 2013

1. Introduction

1.1 Definitions and abbreviations

CA: Companion Animal

CA Project: any IFAW-supported Companion Animal project in which staff working for, or volunteering for, IFAW are responsible for the veterinary activities.

IFAW: International Fund for Animal Welfare

IFAW-qualified veterinarian: a veterinarian with a degree from an accredited veterinary college, who meets local licensing or official authorization requirements, whose clinical skills meet international standards, and who is contracted or employed by IFAW for veterinary responsibilities.

IFAW-qualified veterinary assistant: a person deemed by an IFAW-qualified veterinarian to have been trained to be able to competently perform the duties of veterinary assistant according to the needs of the position. These duties may include a range of responsibilities, including, for example, humane animal handling, anesthetic monitoring, drug calculations, administering injections, applying bandages, animal husbandry, and surgical assistance.

Sterilization Field Manual: the IFAW Field Manual of Veterinary Standards for Dog & Cat Sterilization (this document) and the professional standards to which reference is made therein.

Guardian: person who assumes responsibility for the care of the animal. This may be the animal's owner, rescuer, informal guardian or foster caretaker.

1.2 Basis for the IFAW Sterilization Field Manual

1.2.1. This document outlines basic standards for the surgical, anesthetic and nursing procedures pertaining to sterilization (spay and neuter) of cats and dogs. It is written with a mind to field conditions and the practical considerations of working in the suboptimal environments that field projects often require.

1.2.2. The standards outlined in the Sterilization Field Manual take into account the often challenging field conditions in which we work, and are based on the extensive experience of IFAW staff and their international colleagues in field projects. The guidelines set forth in these documents are not meant to be restrictive, but to assist veterinary staff in prioritizing resources and to ensure consistent and at least minimal international standards.

1.2.3. The animal welfare considerations discussed in Section 2: Patient stress management and animal welfare are implicit in all IFAW veterinary documents, including this one.

1.3 Primary Veterinary Health Care (PVHC)

1.3.1. This document does not include comprehensive PVHC standards. PVHC includes a multitude of veterinary procedures and services, which may vary in emphasis among regions.

1.3.2 PVHC includes:

- issues that pertain to all aspects of animal welfare
- vaccination
- parasite management
- treatment of a variety of emergency medical and surgical conditions
- nutrition
- treatment of traumatic injuries
- infectious and non-infectious disease prevention
- treatment of common diseases
- management of behavioral abnormalities
- biosecurity
- public health considerations

1.4. The IFAW Sterilization Field Manual as a teaching tool

IFAW veterinarians must keep in mind that an important element of most of our projects is to build local capacity. Therefore, it is important to recognize that everything we do as veterinarians and in every interaction that we share with an animal, we teach, we set examples, we establish images and perceptions among the veterinary staff, students and the public who observe us. As such, our actions are likely to influence the future behaviors of trainees even more strongly than our words will. "Do as I say and not as I do" counters the way that people (and animals) learn. It is imperative to work under the principle that people "Do as I do, regardless of what I say", and to be aware that we are always teaching, intentionally or not.

Even if we might sometimes take small shortcuts in a well-controlled clinic with expertly-trained staff because we know from experience that it will not compromise the animal, we must be aware that while we are teaching and serving as role models, we must be consistent, establish routines, and teach through repetition of standards.

Subjective qualification of those standards is a luxury of experience, and once our students gain such experience, they will be able to afford such amendments of their own. But at the outset, it is important to work as consistently as possible within the variability that is inherent to biological systems. To this end, the IFAW Sterilization Field Manual, like other IFAW veterinary documents, is intended to standardize how we teach in our international projects, and to facilitate the conveyance of consistent messages by multiple teachers participating in a given project.

Given the subjective skills that are inherent to excellence in the veterinary profession, it is impossible to write an objective guideline for everything that we do, even for procedures that are performed as often as sterilization of dogs and cats. The documents are not appropriate to be used as a self-teaching manual any more than is a textbook in surgery or pathology.

1.5 Application of the IFAW Sterilization Field Manual

The IFAW Sterilization Field Manual is intended to be used in two ways:

1.5.1. As a guideline for IFAW veterinarians to ensure that everyone maintains and teaches standard protocols for dog & cat sterilization across IFAW projects.

1.5.2. Distributed in hard copy to IFAW grantee field project personnel once these personnel have been trained by an IFAW veterinarian to be able to use the document responsibly, or when they have been evaluated by an IFAW veterinarian and determined to be qualified to use the document responsibly.

1.6 Requirements for use of the IFAW Sterilization Field Manual

1.6.1. All veterinarians and support staff assuming patient management for the CA Project will be expected to practice competently in accordance with the Sterilization Field Manual.

1.6.2. All project veterinary staff must be familiar with this document in its entirety, before project work commences.

1.6.3. Clauses that contain the word 'must' or 'will' are to be prioritized and implemented without exception.

1.6.4. Clauses that contain the word 'should' or 'may' are highly encouraged. These clauses accommodate variability in staff training, experience and preference; the local environment; availability of resources; and other constraints. Staff are expected to use their discretion and to select the most appropriate available procedure or equipment to ensure that animals who are subjected to medical or surgical intervention are at minimum risk of complications.

1.6.5. Questions or concerns regarding application of the Sterilization Field Manual should be directed to the IFAW CA Program Director (or his or her delegate) and/or to the senior IFAW Veterinary Advisor.

1.7. Disclaimer

While IFAW endeavors to train veterinary personnel to international standards of veterinary practice in projects with which IFAW is affiliated, and to train these personnel to use the Sterilization Field Manual responsibly, IFAW is not responsible for, and will not be held liable for, misconduct, accidents, errors, omissions or any undesirable outcomes that occur in the use of this Sterilization Field Manual. Nor shall IFAW be responsible or be held liable for inaccuracies and errors in this Sterilization Field Manual, which is provided "as is" without any warranties of any kind, whether express or implied. The Sterilization Field Manual is intended to be used just like a text book or any other reference publication in the veterinary literature, for which the user is expected to have sufficient training and skills to responsibly implement procedures described therein.

1.8 Appendices and cross-references

1.8.1. The appendices provide extensive notes that are useful for reference as well as for teaching. These are to be used as supplementary material and as a bridge to more detailed information in published references.

1.8.2. Sections are cross-referenced within the body of the document and to Appendices. The links can be followed automatically in the digital version of the document by clicking on the name of the citation.

2. Patient stress management and animal welfare in veterinary care

Appropriate and timely veterinary care is one of the fundamental welfare requirements for animals who are dependent on human guardianship. Within the provision of veterinary care, great attention must be given to minimizing the stress that an animal suffers. Stress over which an animal has no control is severely detrimental to the emotional well-being of the individual, and compromises all the processes that we endeavor to support with veterinary care: healing, recovery from illness, and physiological stability.

Even acute and short-term stress profoundly affects numerous physiologic systems. Blunted activation of the hypothalamic-pituitary axis compromises the many vital systems dependent on it. Suppression of both humoral and cellular immune systems delays healing, interferes with recovery from illness, and increases the susceptibility of an animal to pathogens. Current illnesses may be exaggerated, sometimes to critical degrees. Quality of anesthesia is significantly reduced when an animal is stressed, and may predispose a patient to dangerous complications. These are all issues of which we must be particularly aware in field surgery situations. Management of patient stress is as important as every other element of pre-surgical, surgical and post-surgical care.

In the process of administering veterinary care, we must be aware of the many stressors that we may add to an animal who may already feel unwell. By nature of requiring veterinary treatment, animals are already suffering. They may be ill or in pain. They are often frightened and confused. Their ability to control the responses of their bodies and minds to environmental challenges may be compromised. They suffer metabolic and emotional stress to heal and to fight disease.

Whether animals are ill or undergoing routine sterilization surgery, we must be aware that they are being subjected to a great deal of human handling to which they may not be accustomed, or which they may associate from previous experience to be punitive. When we catch animals for sterilization projects, they are frightened, confused and severely stressed, as anyone would feel on being trapped in a cage or a net and carried off. In a clinic, there are smells of blood, illness, stress and chemicals. There is a great deal of human activity. Lights may be bright, fluorescent, and on at unnatural times. There are sounds of machines, doors, human voices, distressed animals. Animals are confined to cages and forced to submit to our schedules and imperatives. All of these, particularly the latter, compromise one of the most critical elements of the welfare of any living being: the element of choice.

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Personnel who handle animals, particularly fractious or highly anxious animals, must be skilled and efficient in animal restraint. There is never a need to strangle an animal or to stress him or her to the point that she or he is urinating and defecating and screaming in terror. Minimize the number of people involved: the more people crowd around an animal, the more stressed the animal will feel. Speak softly and steadily to the animal, even and particularly when s/he protests. Make a physical examination feel more like a cuddle than being poked and squeezed and pulled and stared at. Approach animals in a way that they feel less threatened. There should be no shouting, slamming of doors, blaring of horns, loud music, cigarette smoke. Never raise your voice at an animal and never strike an animal, as these are not only stressful and potentially cruel, but are most likely counterproductive to whatever you are trying to get the animal to do or stop doing. Cats are usually calmer with their heads covered. Bundle a fearful dog into a blanket rather than choking him on the end of a lead.

Be sure that all supplies that will be needed for the procedure are laid out in advance. A great deal of prolonged stress is caused when staff rush around looking for this or that while the animal is held under restraint. Diagnostic and treatment procedures must be done efficiently and with adequate restraint or sedation to avoid undue discomfort.

A moment's effort of rewarding a patient with a small treat or a cuddle following restraint or other temporary discomfort will greatly improve the tractability and reduce the stress of handling the animal the next time.

Adequate control of pain is mandatory for every veterinary procedure. Good guidelines are now available for assessing pain in animals, as summarized in **Appendix 6: Assessing the need for post-operative analgesia**. By default, if the animal's illness or a procedure done to an animal would be painful to a person, it must be assumed that it is painful to the animal as well, and must be prevented and treated. Post-operative analgesia for common procedures such as spay and neuter are guided by international standards, as outlined in this document.

Analgesia is also an essential component of anesthesia for all procedures that do, or may, cause pain. Sedatives and other drugs that inhibit an animal's physical control over his or her body must simultaneously address the anxiety that is inherent to such a loss of control (e.g., with acepromazine). Anesthesia must be carried out in a way to minimize duration of anesthesia, optimize quality of the effects that anesthesia is meant to induce – e.g., analgesia, muscle relaxation, unconsciousness – and to minimize risks to the animal.

Prior to and following anesthesia, animals must be kept as calm and quiet as possible. Cages should be covered, newly-trapped animals must be allowed to calm down, and human and animal activity, light and noise around the cages must be minimal. All of these factors may not always be possible to control under field conditions. But the stressful effects of many of them can be mitigated through the efforts and innovation of the personnel who care for the animals.

Animals must be anesthetized only when it is certain that the veterinarian will be ready for him or her immediately following induction and preparation for the procedure. Wait times because animals are anesthetized too early are a common cause of unnecessarily long anesthesia.

Personnel must be well informed about what to expect with certain anesthetic drugs, and to monitor the patient accordingly. Emergency intervention protocols must be practiced, and all necessary drugs and equipment in good working order and readily to hand.

Hydration is essential for physiologic stability, healing and well-being. Hydration is inexpensive, and must never be compromised perioperatively or in the treatment of sick animals.

Maintenance of routines has been shown to help animals to cope in a clinic environment. Feeding, cleaning, daily medication and dog walks done at the same time each day appear to return an element of control to animals through the ability at least to anticipate some of the changes to which they are subjected. It is essential also that personnel who feed and clean and walk animals are gentle, move steadily, avoid sudden movements and noise, speak gently to the animals, and understand basic animal communication, e.g., to avoid direct stares and how to approach an animal who is fearful in the cage. This is important not only to reduce the stress of animals, but for the safety of personnel as well.

In addition to the fundamental parameters of cage size, biosecurity, bedding, etc. that are minimum standards for caged animals, additional elements can make a great deal of difference to the stress that an animal experiences while under veterinary care. All cats must have some form of litter pan in the cage – even if it only a plate with shredded paper inside. Dogs must be taken outside to toilet on a regular schedule. If the dog is too ill for this, it is essential to keep the animal clean and to remove waste from the cage immediately. Dogs and cats who are accustomed to living in the street are capable of holding their urine and faeces for days at a time if not taken outdoors, with sometimes grave consequences. Cages and kennels must be kept dry and free of fumes from disinfectants, sewage, vehicle exhaust and other noxious substances.

Retreat space – the opportunity for an animal to remove him or herself from the proximity of people or other animals – is often compromised by the necessities of caging in a clinic or sterilization project. Covering cages with a cloth and placing cardboard or other barriers between them may be essential. Cats should have a small box in the cage in or behind which to hide. Prey species such as rabbits, guinea pigs and hamsters must not be kept in the same areas as predator species such as cats and dogs. These are things that are easy to do, are inexpensive, and can make a great amount of difference to the patient – and therefore also to the effectiveness of the veterinary care.

Temperature is sometimes difficult to control under field conditions. Indigenous animals may be accustomed to high or low temperatures that are common to their environment. It is essential to keep in mind, however, that animals under veterinary care are often unnaturally stressed, have no choice in relocating themselves to an area of more suitable temperature, and may be compromised in their thermoregulatory capacities by illness or anesthesia. All efforts must be made to moderate the temperature and ventilation to a safe and comfortable range for patients.

The examples listed above are by no means exhaustive, but outline some of the common compromises to animal welfare that may be avoided in clinical situations, particularly in field conditions and makeshift clinics such as during a rescue operation. We cannot avoid handling animals, or temporarily to cage them, or to give them drugs that may make them feel woozy, or to subject them to stressful procedures. But we can do a great deal to mitigate the stress that animals suffer while they are under our care. In every direct or indirect interaction with an animal, it is important to be empathetically and cognitively aware of the stressors that he or she may be suffering, to anticipate and preempt them, to relieve them as well as possible, and to de-escalate fear and anxiety. Even if animals are under veterinary care for only a day or two, or for only a few hours as in trap-neuter-release programs, or for a moment to vaccinate, minimization of stress and addressing welfare needs of patients are paramount to professional veterinary practice and to the recovery of animals from surgery or illness.

Under no circumstances may we compromise welfare standards with the rationalization that animals are "stray" or "are used to a hard life". Animals are not lesser beings for lacking adequate human guardianship, nor do they suffer less. Animals must never be subjected to substandard welfare during veterinary procedures or for research, or to unnecessary risks for students to practice skills. It is our responsibility and privilege as veterinarians and as welfare guardians to ensure that all animals are held in equal respect.

3. Management of IFAW field veterinary projects

3.1. Veterinary staff qualifications and responsibilities

3.1.1. An IFAW-qualified veterinarian must be assigned to every field project that is under IFAW's jurisdiction and that involves animal health, in order to ensure that the requirements outlined in the Sterilization Field Manual are being met.

3.1.2. All team members must respect final decisions on veterinary care made by the veterinarian in charge in the project.

3.1.3. The veterinarian in charge, together with the IFAW CA Program Director (or his/her designee), must determine the level of responsibility that veterinary staff and volunteers are qualified to assume within any specific project. Assignments must be in accordance with the existing guidelines of the Sterilization Field Manual, local law and general veterinary practice.

3.1.4. Veterinary staff must meet the qualifications outlined in Section 1.1 Definitions and abbreviations. Project veterinarians and veterinary assistants with insufficient qualifications or experience must undergo additional supervised training and evaluation by IFAW-qualified veterinarians. A subsequent evaluation of the veterinarian's or assistant's competence will be made by an IFAW-qualified veterinarian before the person may work independently.

3.1.5. Volunteer veterinary staff must follow the requirements and rules of the IFAW Volunteer Veterinary Staff guideline. It is strongly recommended that all volunteers be asked to sign a document similar to that shown in Appendix 16: Legal forms for IFAW project participants in order to ensure that they are properly informed of risks and responsibilities, and to protect the project organizers in the event that something goes wrong.

3.2. Compliance with local regulations and legal requirements

3.2.1. CA Project activities must comply with all legal requirements for the operation of a veterinary facility in the locality, including:

- staff qualifications
- permits and licenses
- recording and monitoring systems for controlled drugs, anesthetic equipment, x-ray equipment and any other equipment or substances that may pose a health risk to staff.
- incidents and accidents
- disposal of animal bodies, clinical waste (organic matter), and sharps
- use and location of premises

3.2.2. In the event that legally-allowed drugs are inadequate in a given location to ensure the optimal safety and health of a patient, alternative drugs may be used with the permission of local collaborators, and according to veterinary practice standards governing the use of such drugs in the United States, European Union or Australia.

3.2.3. Where animals are subjected to anesthesia, surgery, hospitalization or euthanasia, guardians must sign forms agreeing to the procedures. In countries where it is relevant to do so, guardians should sign their consent to the off-label use of drugs. Sample forms may be found in Appendix 16: Legal forms for IFAW project participants.

3.3. Preparation for accidental exposure of staff to dangerous drugs or pathogens

3.3.1. For expedient treatment of a person accidentally exposed to veterinary drugs, drug information data sheets and other information sheets for all drugs and vaccines used in a CA project should be current and readily accessible by all staff.

3.3.2. Necessary antidotes and a first aid kit should be readily available and accompany a staff member to the local human hospital for administration if the need arises. Human treatment clinics in some regions may not have necessary antidotes available, and it is best to have them ready to accompany the patient. Information on the use of antidotes and other relevant information must be included in the emergency kit.

3.3.3. All personnel who are handling animals are advised to be fully vaccinated against all locally-relevant zoonotic pathogens (including rabies) and endemic preventable diseases; refer to WHO and CDC web sites for country-specific vaccination guidelines: http://www.who.int/ith/en or http://wwwnc.cdc.gov/travel/destinations/list.htm with an internationally recognized vaccine (e.g., listed by the WHO).

3.3.4. Employers and staff managers must be aware that in some countries (e.g., United States) one cannot mandate staff to be vaccinated or to take other kinds of prophylactic treatment against endemic pathogens. One may only advise staff of potential risks and provide guidelines for prophylactic care. For IFAW projects, personnel must sign a waiver of responsibility to hold IFAW harmless in the event of illness (Appendix 16: Legal forms for IFAW project participants). Other project owners are encouraged to provide personnel with similar guidelines and waivers of responsibility.

3.4. Records and animal identification

3.4.1. All animals who receive veterinary care through an IFAW CA project must have written records that describe information pertinent to the activity and according to local legal requirements.

3.4.2. All records for IFAW CA projects must include:

- date of activity
- identification of the animal (if there is any)
- basic description of the animal (color, unique markings or characteristics, fur length)
- sex
- estimate of age
- identifying markers such as tattoo or microchip
- name and contact information of guardian or owner, if there is one

3.4.3. Medical and surgical records must include:

- clinical history, if available
- physical examination results
- diagnostic tests, if done
- all drugs – name, dose, route of administration, time of administration
- anesthetic record – minimally this must include record of drugs (name, dose, route of administration, time of administration), complications, emergency drugs administered
- surgical record if the procedure involved anything other than a standard ovariohysterectomy or castration (e.g., as described in Fossum et al. 1997, Small Animal Surgery)

3.4.4. If the animal is retained in a hospital or shelter, the animal should be identified by his or her individual hospital record, together with a corresponding ID on the animal (e.g., numbered collar, tag, microchip, or cage ID for animals who cannot be handled).

3.4.5. Sample record forms may be found in Appendix 14: Examples of clinical forms and record sheets.

4. Pre-surgical evaluation and considerations for stages of the estrous cycle

4.1. Primary veterinary care prior to the sterilization event

4.1.1. If primary health care provision prior to the sterilization event is not possible, the animals must be vaccinated at the time of surgery. Unless animals have recently been dewormed, this should be done at the time of surgery, or oral de-worming medication sent home with the guardian. Refer to Section 9: Vaccination and parasite control.

4.1.2. All dogs and cats must be vaccinated against rabies, either before surgery or at the time of surgery. Previously-vaccinated animals must be current on rabies vaccination status per local laws (i.e., annual or once every 3 years), Section 9: Vaccination and parasite control.

4.1.3. It is strongly recommended that dogs and cats be vaccinated with the core vaccines in addition to rabies either before surgery or at the time of surgery. A schedule of core vaccines may be found in Appendix 3: Vaccination Guidelines.

4.2. All animals must undergo a standard physical examination on the day of surgery. This examination should minimally include assessment of all major organ systems according to the physical examination guideline in Appendix 14: Examples of clinical forms and record sheets. Where possible, a health history should be obtained from the animal's guardian as well.

4.3. In areas in which certain diseases are endemic that may compromise the patient's survival under anesthesia or recovery from routine surgery, additional diagnostic tests may be performed prior to surgery wherever possible (e.g., for heartworm or babesiosis).

4.4. In some extreme instances, when the animal is too aggressive and dangerous to examine safely prior to anesthesia, he or she may be tranquilized first, as long as no major abnormality is observed (e.g., significant neurologic dysfunction, large tumors, etc.). A physical examination must be done as soon as the animal can be handled safely and before he or she is subjected to surgery.

4.5. An animal must not be subjected to anesthesia and/ or any surgical procedure if the physical examination and/or medical history suggest any undue or unnecessary risk for an elective surgery.

4.6. When the physical examination presents evidence of disease, injury or malnutrition that is likely to compromise the patient's ability to withstand or recover from anesthesia and surgery:

4.6.1. The animal must be evaluated and treated according to the guardian's agreement, available resources, and welfare prognosis for the animal.

4.6.2. When clinical history or findings suggest that the animal may risk spreading infectious disease to other animals or to people (e.g., distemper, parvo, rabies, kennel cough), s/he must be handled first and foremost according to human and animal biosecurity standards (e.g., in case of rabies). Treatment, isolation and other options depend on available resources.

4.6.3. Humane euthanasia may need to be considered in certain situations: refer to Section 10: Euthanasia.

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4.7. Female dogs and cats may be spayed at any stage during the reproductive cycle. Additional caution must be used when sterilizing bitches or queens who are in estrus or who are pregnant.

4.7.1. The surgeon must be aware that tissues may be more friable and perfusion to the reproductive tissues increased, and take appropriate precautions.

4.7.2. Intravenous fluid therapy may be necessary. An IV catheter should be placed to maintain hydration and blood pressure throughout surgery, and to allow rapid initiation of fluid therapy if necessary. Refer to Section 7.3: Intravenous fluid support during surgery.

4.7.3. Ideally, and if the option is reasonable, avoid spaying a bitch within 8 weeks after estrus. This is the luteal phase of the estrous cycle, and spaying during this time will cause a precipitous decrease in progesterone, which may induce clinical signs of false pregnancy (pseudopregnancy or pseudocyesis). However, if it is unlikely that there will be another opportunity to spay the bitch, this risk is considered acceptable in favor of ensuring that she is spayed promptly and safely.

NB: a common myth suggests that spaying during pseudocyesis will prolong the clinical signs of false pregnancy. This is incorrect. The clinical signs begin with regression of the corpus luteum and the consequent drop in progesterone production. Removal of the corpus luteum by ovariohysterectomy will not worsen the condition.

4.7.4. Dogs and cats with pyometra must be spayed immediately, as an emergency surgery. Surgery must be preceded and followed by appropriate antibiotics and supportive care.

4.8. Post-partum bitches and queens

4.8.1. It is preferable that bitches and queens are not spayed during the first 6 weeks post-partum. This guideline is out of concern for dependent neonates and to give the uterine tissues and associated vasculature time to recover. However, if it is not possible to ensure that the bitch or queen will be spayed if surgery is delayed, then she may be spayed during lactation, providing that her offspring are not compromised as a result.

4.8.2. Puppies and kittens who are dependent on their mother's milk must not be away from the mother for more than 4 hours. The mother's surgery must be planned in a way to ensure that her total time away from the pups is minimized. The neonates must be kept warm, safe and fed (if necessary) while separated from their mother.

4.8.3. The lactating bitch or queen should not be starved for more than 8 hours prior to surgery. Water must always be present and must never be withheld. Full-time access to drinking water is particularly critical for a lactating female, as her water requirements are very high.

4.8.4. Mother and offspring should be closely monitored post-operatively to ensure that the puppies or kittens are not accidentally injured or in case the mother has trouble resuming maternal care.

4.8.5. Post-partum queens should be spayed by the flank technique, unless there is reason to suspect uterine pathology. This reduces risk of milk leakage into the abdominal incision, and allows the kittens to continue to nurse with less trauma to the surgery site than following a midline incision.

4.8.6. Refer to Appendix 5: Anesthetic and analgesic drug protocols for recommended anesthetic protocols for lactating bitches and queens.

4.9. It is recommended that dogs and cats are sterilized prior to puberty whenever possible. This is recommended on grounds of medical, behavioral and population management considerations. (Note that healthy kittens may become pregnant as early as 4 months!) For puppies and kittens younger than 16 weeks of age, refer to Section 8: Early-Age Sterilization.

5. General clinical considerations

5.1. A new, sterile, disposable needle must be used for parenteral administration of any drug or vaccine.

5.2. If necessary, syringes may be re-used if:

5.2.1. The syringe is not contaminated with any bodily fluids from an animal.

5.2.2. The syringe tip is always protected with a capped needle. If a syringe is left uncapped, it must be discarded (or sterilized before re-use).

5.3. Syringes may be washed and autoclaved to re-use them. All contents must be completely washed out with water and a pipe cleaner or test tube brush. The syringe must be rinsed well with clean water and then packaged so that sterility is maintained after autoclaving, until use.

5.4. Do not use chemical sterilization ("cold sterilization") for syringes.

5.5. All medical supplies, including drugs, vaccines and other consumables must be stored according to the manufacturer's recommendations.

5.6. Use of expired drugs and supplies

5.6.1. All medical supplies, including drugs, suture materials, vaccines, sterile materials and other products with expiry dates, should not be used if they are 6 months beyond the manufacturer's expiry date, or if there is any evidence of breach of sterility, contamination, or reduction in effectiveness or safety (e.g., from exposure to undue heat, cold or sunlight).

5.6.2. The decision to use a product beyond its expiry date rests with the head veterinarian for the project.

5.6.3. If there is any doubt regarding the expired product's safety or effectiveness, it should not be used.

5.7. When consumable products such as intravenous fluid bags or bottles and tubes containing medicine are opened, the date of their opening should be clearly marked on the package.

5.8. Opened products should be discarded if they have been contaminated, or at the head veterinarian's discretion. Products must be discarded according to manufacturer's instructions and in a way that meets local laws.

5.9. All containers (including syringes) containing a drug or vaccine must be clearly labeled with the contents (name of drug & concentration).

5.10. All drugs that are given to animals must be calculated based on the patient's body weight. Body weight should be measured, rather than estimated, whenever possible.

6. Anesthesia

6.1 General considerations and preparation for anesthesia

6.1.1. If the physical examination or medical history of the animal suggest that surgery may cause more than regular risk to the patient, elective surgery such as sterilization should be delayed. Refer to **Section 4: Pre-surgical evaluation and considerations for stages of the estrous cycle**.

6.1.2. Supplies for treating anesthetic complications must be completely prepared and easily available before the animal is anesthetized. All veterinary and nursing staff must be thoroughly familiar with emergency protocols and location of supplies. Refer to **Appendix 9: Emergency drugs quick reference drug chart**, **Appendix 10: Emergency treatment kits**, **Appendix 11: Emergency treatment of cardiac and respiratory arrest** and **Appendix 12: Emergency treatment for anaphylactic reaction**.

6.1.3. All possible measures must be taken to minimize stress and anxiety in patients prior to anesthesia. Stress not only compromises physiologic processes necessary for homeostasis and healing, but may severely compromise the safety and quality of anesthesia. Refer to **Section 6.7: Induction and recovery from anesthesia**.

1) Litters of puppies or kittens, and animals who are familiar and comfortable with one another should be kept together as long as possible.

2) Cages should be covered with cloths or cardboard to reduce visual stimulation.

3) Cages for cats should contain a small cardboard box or other area into which cats can withdraw or hide.

4) Animals awaiting surgery should be kept away from the admission and recovery areas. Admission areas are full of anxious, excited animals and people. This environment is much too stressful and stimulating for animals recovering from anesthesia. Recovery areas are full of animals who behave abnormally, smell abnormally and make abnormal sounds. This is immeasurably distressing to un-anaesthetized animals who observe them.

5) The environment for animals awaiting, or recovering from, anesthesia should be as quiet as possible. If possible, the areas should be dimly lit. There must be no shouting, loud doors, banging cages, smoking, music or other avoidable disturbances.

6) Animals must be handled and restrained as gently as possible. This is challenging sometimes when animals are fractious or particularly fearful. A great deal can be achieved with soft tone of voice, slow and gentle approach, steady movements that do not startle the animal, non-threatening body language, and practiced animal handling skills.

7) Cats usually calm down markedly if the head is covered or if they can hide inside of a towel or cloth bag. Dogs usually become more anxious with their heads covered.

8) If an animal is caught in the street or brought to the clinic under other stressful conditions, he or she should be allowed to calm down in a covered cage for at least 30 minutes before induction of anesthesia.

9. Animals who are caught with a net must be moved immediately to a cage to calm down and prepare for anesthesia. If the animal is too fractious to move safely to a cage, he or she may be anaesthetized immediately and then moved, per **Section 4.4.** Animals must never be left in the net under any circumstances.

6.1.4. Fasting

1) Water must be always available to animals. Do not withhold water prior to anesthesia.

2) Animals older than 6 months of age should have food withheld before anesthesia for at least 8 hours but no more than 16 hours.

3) Animals ca. 4-6 months of age should have food withheld for 4-8 hours (e.g., feed at 8 AM, do surgery in the afternoon)

4) Animals less than 16 weeks of age should have food withheld for 30-60 minutes prior to anesthesia. Small meals may be fed for up to 30 minutes prior to anesthesia. Do not fast for more than 1 hour. Refer to Section 8, Early-Age Sterilization.

5) If it is likely that the animal has been fasting too long (according to the guideline above), he or she should be fed immediately and surgery delayed until an appropriate time later. Hypoglycemia is a serious risk, particularly for young animals.

6) Animal guardians who bring dogs and cats for sterilization must be informed in advance of fasting times for the animal.

7) Veterinary staff must be prepared that dogs and cats often will have food in their stomachs despite instructions to guardians for fasting or if the animal was trapped from the street. Emesis should be expected and perhaps purposely induced with pre-anesthetic drugs.

6.1.5. Anesthetic drug doses should be calculated on the basis of measured body weight. Estimated body weights are sometimes used in the event of an emergency, but this should not be done as a matter of routine.

6.1.6. It is imperative that anesthesia time be minimized. Calculate <u>all</u> drug doses and prepare <u>all</u> drugs, supplies and equipment <u>before</u> the patient is anesthetized.

6.1.7. An anesthetic record must be kept for each patient according to specifications outlined in Section 3.4: Records and animal identification and Appendix 14: Examples of clinical forms and record sheets.

6.1.8. If gas anesthesia is used, the room must have adequate ventilation. A scavenging system or waste gas escape system must be in place to protect the patient and staff.

6.1.9. Prior to anesthetic induction, the animal should be encouraged to empty his or her bladder and bowels. Dogs may be taken on a short walk to encourage this. With cats it may be difficult to achieve this, particularly if they feel stressed.

6.2 Intravenous catheters

6.2.1. All animals should have a new, sterile IV catheter placed just before or immediately after induction of anesthesia. This allows rapid administration of emergency drugs, administration of intravenous fluids, and re-dosing of anesthetic drugs.

6.2.2. All bitches and queens who are more than 4-6 weeks pregnant must have a new, sterile, intravenous catheter placed prior to surgery. For bitches an 18-22G intravenous catheter, and for queens a 22-24G intravenous catheter are usually appropriate.

6.2.3. All pediatric patients undergoing early-age sterilization must have an IV catheter (Section 8, Early-Age Sterilization).

6.2.4. To place an IV catheter, the fur must be clipped over the vein (cephalic or saphenous vein) to allow easy visibility of the vein and to avoid adherence of fur to the sterile catheter. The venipuncture site must be cleaned with 2-4% chlorhexidine or povidone iodine/alcohol (Appendix 15: Disinfectants) before the catheter is placed.

6.3. Endotracheal intubation

6.3.1. All animals must be intubated immediately after induction of anesthesia (regardless of whether or not the animal will be maintained on gas anesthesia), and must remain intubated until they are able to control their own airways again (swallowing, coughing).

6.3.2. Endotracheal intubation must be performed with care and skill to avoid trauma to the trachea or intubation of the esophagus. Cats are particularly vulnerable to tracheal trauma. Lidocaine must be used to intubate cats (see below).

6.3.3. All endotracheal tubes must be checked prior to each use for patency and an effective cuff.

6.3.4. The endotracheal tubes must be thoroughly cleaned after each patient according to the following four steps. Do not use tubes in subsequent patients until all steps have been completed.

1) Wash the tube thoroughly and clean the inside with a pipe cleaner or test tube brush to remove all mucus and other soiling.

2) Soak in disinfectant for 15 minutes (see Appendix 15: Disinfectants for appropriate disinfectant options).

3) Rinse thoroughly with drinking water. This is very important to avoid introduction of chemical disinfectant into the airways of the next patient.

4) Air-dry or shake off as much water as possible before use in the next patient.

6.3.5. Endotracheal tubes must be lubricated with a sterile, water-soluble lubricant prior to placement in the patient's airway.

1) The lubricant must be free of dyes, perfumes and spermicidal chemicals. Non-spermicidal 'personal' lubricants (e.g., K-Y Jelly) are a good choice.

2) Do not use oil-based lubricants (e.g., eye ointment or wound salve).

6.3.6. Topical laryngeal anesthesia prior to intubation must be done in cats (and is helpful in some dogs as well).

1) Lidocaine is sprayed onto the back of the pharynx, onto the glottis. Wait 1-2 minutes, then intubate.

2) Lidocaine dose

- Draw up 0.1-0.3 ml of 2% lidocaine solution in a 1-ml syringe. For cats < 2 kg, use 0.1-0.2 ml. For cats 3 or more kg, use 0.2 – 0.3 ml.
- Remove the needle and spray the lidocaine directly from the syringe.
- Alternatively, use the commercial pediatric lidocaine spray.
- The maximum safe dose of lidocaine is 3-4 mg/kg. Do not use more than one dose.

3) Refer to Appendix 5: Anesthetic and analgesic drug protocols

6.3.7. The use of a laryngoscope greatly aids in the efficiency of intubation, particularly in cats and brachycephalic dogs.

6.3.8. Never force an endotracheal tube into the trachea. If it doesn't enter easily, then something is wrong. If patients frequently develop laryngospasm, sore throats, or the endotracheal tube has blood on it when it is removed, the intubation technique must be reviewed and re-learned. These are common problems caused by inappropriate intubation methods.

6.3.9. Endotracheal tubes may remain un-cuffed if the tube is not used for delivery of anesthetic gas. This is particularly important in cats, as the feline trachea is so easily traumatized. Be sure that the cuff is not over-inflated.

6.3.10. Remember to deflate the cuff before the tube is removed. The trachea can sustain great damage if a tube is removed while cuffed.

6.4. Eye lubrication

6.4.1. The eyes must be lubricated with a sterile ophthalmic lubricant as soon as possible following induction and intubation. This is particularly important in animals anesthetized with ketamine and in all cats.

6.4.2. The lubricant should be a preparation of "artificial tears" (ointment, not drops). These usually contain hydroxypropyl methylcellulose, lanolin, polyvinyl alcohol or carbomer. The lubricant should not contain antibiotic. However, if antibiotic eye ointment is the only option available, it may be used.

6.4.3. Avoid applying the ointment directly onto the cornea. Rather, form a small pocket with the upper or lower eyelid by drawing it gently away from the eyeball. Instill the ointment into the conjunctival pocket, then close the eyelids and gently massage the ointment onto the eye with the eyelids closed.

6.4.4. Avoid touching the tip of the tube to the conjunctiva, so that pathogens are not transmitted from one patient to another.

6.5 Prevention of hypothermia

6.5.1. Care must be taken to prevent hypothermia (rectal temperature less than 37°C).

6.5.2. Hypothermia is a common complication in anesthesia, especially in cats, small dogs and immature animals. It may delay recovery from anesthesia and cause other, sometimes life-threatening, complications.

6.5.3. Refer to Section 8: Early-Age Sterilization for information on preventing and managing hypothermia and hypoglycemia in pediatric patients.

6.5.4. All animals should receive supplemental heat if ambient temperatures are below 25°C. Very small or immature animals may require supplemental warmth at even warmer ambient temperatures.

6.5.5. Supplemental heat sources may be provided in the form of electric heat pads, microwavable liquid heat pads, warm water bottles, or socks filled with (uncooked) rice, barley, lentils or other small grains or legumes and heated in the microwave.

6.5.6. Care must be taken that the heat source is not so hot that it will scorch the skin. Heat sources should be approximately 38°C. Check the temperature as you would a baby's bottle: hold it against your inner wrist. It should feel pleasantly warm. If you feel the need to remove your hand within 60 seconds, then it is too hot. If it feels cooler than your skin, then it is too cold.

6.5.7. A towel or blanket must always be placed between the heat source and the animal's skin.

6.5.8. If the animal begins to shiver or rectal temperature falls below 37°C, supplemental heat must be provided to raise body temperature. Patients must be supervised until they are stabilized. If the body temperature fails to recover, it may be necessary to administer warm fluids intravenously. Refer to Appendix 7: Vital parameters during anesthesia.

6.6 Anesthetic monitoring

6.6.1. Refer to Appendix 7: Vital parameters during anesthesia and Appendix 8: Guide for monitoring depth of anesthesia for guidelines on anesthetic monitoring.

6.6.2. The 5 stages of anesthesia include:

1) Pre-anesthetic (or pre-induction) period
2) Induction
3) Maintenance
4) Recovery
6) Post-anesthetic period

6.6.3. Minimum monitoring parameters must include:

1) heart rate & rhythm
2) perfusion (capillary refill time)
3) peripheral pulse: strength and rate
4) respiratory rate and quality
5) plane of anesthesia
6) rectal temperature

6.6.4. Parameters 1-5 must be monitored at least every 5 minutes from induction through recovery.

6.6.5. Rectal temperature must be measured every 5 minutes in pediatric patients (cf. Section 8: Early-Age Sterilization), every 10-15 minutes in euthermic, healthy adults, and every 5 minutes in adults whose body temperature is abnormal or nearly abnormal.

6.6.6. A stethoscope must be used for the anesthetic monitoring of patients. Pulse oximeters, capnographs, blood pressure cuffs and other instruments are useful as supporting tools but must never be used in place of a trained person actively monitoring the patient. Do not rely on the beep of a machine as the sole method of monitoring.

6.6.7. The anesthetist must be familiar with the drugs and the effects that they are likely to produce. See Appendix 5: Anesthetic and analgesic drug protocols.

6.6.8. Emergency drugs must be readily available and properly stocked for rapid access See Appendix 10: Emergency treatment kits.

6.6.9. The patient must be in a surgical plane of anesthesia throughout the surgical procedure. See Appendix 8: Guide for monitoring depth of anesthesia.

6.6.10. Animals must never be left unobserved while intubated.

6.7 Induction and recovery from anesthesia

6.7.1. During induction and recovery from anesthesia, it is imperative to minimize stimulation of the patient.

1) In Plane 2 of anesthesia during induction and recovery (Appendix 8: Guide for monitoring depth of anesthesia), animals may be hyper-excitable but do not have normal motor control. Moreover, the gag reflex may not be fully functional. Reaction to stimuli during this transition puts the animal in danger of injuring him or herself or someone else, and of compromising his or her respiration.
2) Any stimulation during induction will reduce the effectiveness of anesthetic drugs and the quality of anesthesia. The more quiet and relaxed the animal is before and during induction, the less anesthetic needs to be used. This saves costs to the project and risk to the animal.

6.7.2. The patient must be monitored carefully during induction and recovery to ensure timely intervention in case of emergency. Animals may become malpositioned during these times and risk obstruction of airways or other critical situations.

6.7.3. Removal of the endotracheal tube

1) Uncuff the endotracheal tube before removal!
2) Dogs: remove the endotracheal tube when the patient begins to swallow or cough. Other signs that the patient is ready to be extubated include voluntary movement of the limbs or head.
3) Cats often begin to move the head, limbs or tail before they begin to swallow or cough. Remove the endotracheal tube at the first of these signs. Delaying removal of the tube may result in laryngospasm.

6.7.4. Following extubation, the patient must be checked at least every 5 minutes until he or she is able to stand.

6.7.5. As soon as the animal is able to stand:

1) Offer water as soon as there is no risk that the animal will accidentally drown in the water bowl.
2) Young animals (less than 16 weeks old) should receive food and water as soon as possible to avoid hypoglycemia. Food should be soft (e.g., tinned food), easy to consume and easily digestible. These patients may require encouragement with oral or intravenous glucose if they appear weak and unwilling to eat right away.
3) Hypoglycemia is an important concern in pediatric patients. Refer to Section 8.2: Pediatric physiology in anesthesia.

6.7.6. It is essential to keep animals warm during recovery. This is particularly true for Early-Age Sterilization patients. During anesthesia, thermoregulation is compromised, and young animals may quickly become hypothermic. Refer to Section 6.5: Prevention of hypothermia.

6.8 Anesthetic drugs

6.8.1. General anesthesia is defined as a loss of consciousness combined with a loss of sensation. This should include hypnosis, loss of reflexes, analgesia and muscle relaxation. The effect usually requires a combination of drugs.

6.8.2. There are a variety of suitable anesthetic protocols for use in field and clinic sterilization surgeries. Suggested options and considerations may be found in Appendix 5: Anesthetic and analgesic drug protocols.

6.8.3. The anesthetic protocol and its implementation must:

1) produce a surgical plane of anesthesia for a duration long enough for the surgery;
2) present as little risk as possible for causing cardiovascular, respiratory or neurologic crises;
3) produce adequate pain control throughout the most painful parts of the surgery; and
4) allow the use of as little drug as possible while meeting the conditions above.

6.8.4. For anesthetic protocols for early-age sterilization, refer to Section 8: Early-Age Sterilization.

6.8.5. If using gas anesthesia, the vaporizer must be specific for the drug, and all equipment must be installed, calibrated and maintained according to manufacturer specifications.

6.8.6. When using gas anesthesia, the room must have adequate ventilation. A scavenging system or waste-gas escape system must be in place to protect the patient and staff.

6.8.7. Do not anesthetize animals until the surgeon can be ready for them as soon as they have been induced and are prepared for surgery. It is unacceptable to prolong anesthesia only to wait for a surgeon to be ready.

6.9 Post-operative analgesia

6.9.1. Analgesia must be provided to all animals subjected to sterilization surgery, and should extend to at least 24 hours post-surgery.

6.9.2. Analgesic drug options, doses, and considerations for particular drugs may be found in Appendix 5: Anesthetic and analgesic drug protocols. This is usually a non-steroidal anti-inflammatory (NSAID). The first dose of this is given during surgery so that it is in effect by the time the patient wakes up.

6.9.3. The need for further analgesia should be evaluated on a case-by-case basis. Common needs for this include inflammation, infection or other complications with the surgical wound, or as adjunct management of the patient licking, scratching or otherwise traumatizing the wound. Refer to Appendix 6: Assessing the need for post-operative analgesia for detailed guidelines.

6.9.4. Never use corticosteroids to control post-surgical pain or inflammation.

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7. Surgery

7.1 Surgical environment

7.1.1. Field spay/neuter surgeries are often done in makeshift surgery areas. Depending on climate and ambient temperatures, the 'surgical theater' may be a large indoor room in a community building, for example, or a reasonably sheltered outdoor area, e.g., a temple courtyard or large tent.

7.1.2. Outdoor or open-air surgical areas must have reasonable protection from elements, reasonable protection from wind and raised dust (minimize contamination of surgical fields), and ambient temperature that does not compromise the survival of animals who are under the stress of anesthesia and surgery. Ideally, this area has some kind of a roof, whether from a tent or other structure.

7.1.3. The area where surgery is performed must be well ventilated. Ideally, it will be possible to regulate ambient temperature to prevent extremes (maintain between 21°C - 27°C).

7.1.4. The use of air conditioning must be balanced against the risk of blowing dust, fungal spores and other air-borne pollutants in the surgical theatre.

7.1.5. A routinely-used indoor surgical suite must be kept clean at all times, and a cleaning protocol should be written and followed. Elements of a surgery room cleaning protocol include:

1) It is important that there are as few objects as possible in the surgery room. All supplies that are kept there must be stored in cupboards or in plastic bins that are easily wiped down.
2) At the beginning and end of the surgery day, all surfaces in the surgery room must be washed with disinfectant solution. All vertical and horizontal surfaces should be wiped clean (tables, counters, walls, cupboard doors, table legs, floors, windows, equipment, lamps, etc.).
3) If there are air conditioners in the room, these must be professionally cleaned and vacuumed at least once per month; ideally once per week. It is advised that air conditioners are kept off while surgery is in progress.
4) The rubbish bin must be emptied at the end of each day, or more often as it fills.
5) The rubbish bin must be washed at the end of each day and disinfected (even if a plastic bin liner is used).
6) The cleaning equipment, e.g., bucket, mop, cloths, must be used only in the surgery room. These items must not be used in any other area.
7) Disinfectants that are appropriate for use in the surgery on inanimate objects are listed in Appendix 15: Disinfectants.

7.1.6. Between surgical patients, the operating table, surgical instrument tray and immediate surrounding surfaces (including floor) must be washed of all organic material (blood, urine, faeces, fur, saliva, tissue). The table and instrument tray must be wiped with disinfectant before use for the next patient.

7.1.7. Ideally, the patient is prepared for surgery (e.g., fur clipping, IV catheter placement) in a room that is separate from the surgical suite, or at least on a table that is not the surgery table.

7.1.8. Traffic through the surgical area should be kept to a minimum and include only staff who are immediately involved in the surgery or in preparation of the animals for surgery.

7.2 Preparation of the animal for surgery

7.2.1. The urinary bladder should be palpated before the animal's skin is cleaned for surgery. Dogs should be taken outdoors for the opportunity to void bladder and bowels prior to induction of anesthesia, but if animals are anxious, this is not always achievable. If the bladder is found to be full once the anesthesia has been induced, an effort may be made to gently express it manually. This must never be forced and the effort must be abandoned if gentle pressure doesn't result in voiding of urine. If the bladder is so distended as to interfere with surgery, it may be expressed by the surgeon once the abdomen has been opened.

7.2.2. Fur must be removed cleanly down to the skin in the area where the incision will be made.

7.2.3. Fur may be removed with electric clippers (#40 blade) or with a disposable razor.

7.2.4. Chemical depilatory agents must never be used.

7.2.5. Great care must be taken to avoid traumatizing the skin during clipping, as this trauma may encourage the animal to lick or chew excessively after surgery. The most common reason for animals to bother their surgical incisions is because of clipper or shaving wounds on the skin. Even very small clipper wounds or scratches will become itchy and cause the animal to lick.

7.2.6. The clipped area must be long and wide enough to ensure that there is no possibility of fur extending into the surgical window of the draped area.

1) On females undergoing midline ovariohysterectomy, the clipped area should extend from cranial to the umbilicus, caudal to the pubis and lateral to the teats. In long-haired animals, a larger area may need to be clipped.

2) On female cats undergoing a lateral ovariohysterectomy (flank spay), a square of fur at least 5-6cm long and wide should be removed from over the ilium and greater trochanter, extending cranially and ventrally. In long-haired queens, a larger area may need to be clipped.

3) On male dogs, the clip should extend from mid-way on the prepuce to the perineum.

4) The fur of male cats is removed from the scrotum and the immediately-surrounding perineum. Fur may be plucked or clipped; plucking often does less damage to the skin and results in less post-operative licking.

7.2.7. All clipped fur must be completely removed from the animal, from on and around the animal, and from the clothing of personnel who will subsequently enter the surgical suite. This may be done with a small, hand-held vacuum cleaner or sticky-tape.

7.2.8. The skin within the clipped edges must be cleaned with clean cotton wool or gauze swabs and 2- 4% chlorhexidine or dishwashing liquid soap (for sensitive skin) until the skin is clean. Avoid scrubbing the skin, as this causes micro-abrasions which later cause the animal to lick the skin.

7.2.9. Once the skin is cleaned, the surgical area is disinfected. A standard ‘outward’ skin preparation technique must be used, beginning at the center of the clipped area (where the incision will be made) in a circular fashion outward to the edges of the clipped margin. Do not go back in the other direction.

- Clean gauze or cotton wool is used for application of disinfectant. Cotton wool is preferable to gauze swabs, as it is less abrasive.
- Disinfectant may be chlorhexidine 2-4%, or alternating swabs of povidone iodine and 70% isopropyl alcohol, or other disinfectants appropriate for skin preparation (cf. Appendix 15: Disinfectants)
- Alcohol should be avoided on pediatric patients to avoid causing hypothermia.
- The pre-surgical skin disinfection must be done for a minimum of 8 successive repetitions.
- Be as gentle as possible. Micro-abrasions of the skin caused by cleaning will result in the animal licking and scratching at the surgical site after surgery, just as rough clipping will.

7.2.10. Once the skin is prepared (clipped, cleaned and disinfected) for surgery, care must be taken that the surgical site is not touched or otherwise contaminated. If the surgeon will not begin the surgery for more than 5 minutes, a clean gauze swab soaked in skin disinfectant should be placed over the disinfected area to completely cover the anticipated incision site. Immediately before the surgeon begins the incision, the area should be re-sprayed with skin disinfectant. If the delay is longer than 20 minutes, the skin must be disinfected again.

NB: All efforts must be made to avoid anesthetized patients waiting for the surgeon. Prolonged durations of anesthesia due to poorly-timed inductions are not acceptable.

7.2.11. Avoid wetting the animal’s skin or fur excessively, as this may lead to hypothermia.

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7.3. Intravenous fluid support during surgery

7.3.1. Pregnant females must have intravenous fluid support throughout surgery and at least through anesthetic recovery.

7.3.2. Patients who are in any way compromised or considered to be at higher risk than normal must receive intravenous fluids during surgery and until they are stabilized after recovery.

7.3.3. Fluids should be administered to patients whose surgeries take longer than 30 minutes.

7.3.4. Fluid rates before and after surgery are 2-4 ml/kg/hour. During surgery, fluids are administered at 10 ml/kg/hour. It is particularly critical for EAS patients to ensure that fluids are calculated and delivered accurately (Section 8.2: Pediatric physiology in anesthesia).

7.3.5. Appropriate fluid options for physiologically stable surgical patients include 0.9% NaCl (physiologic saline), lactated Ringer’s solution or Hartmann’s solution.

7.3.6. If hypothermia is a risk, it is helpful to warm fluids to 37°C.

7.4. Preparation of the surgeon for surgery

7.4.1. Clean, short-sleeved surgical scrubs or a sterile surgical gown must be worn throughout surgery. Plastic aprons, cleaned or changed between patients, may be used as well. There must not be any fur or other debris on the outer clothing.

7.4.2. The surgeon and surgical assistants should wear a surgical cap to ensure that no human hair contaminates the surgical site. Uncovered hair is a common source of contamination.

7.4.3. The surgeon and surgical assistants should wear a surgical mask during surgery.

7.4.4. A new pair of surgical gloves must be worn for each patient (Section 7.5: Surgical gloves and drapes)

7.4.5. Surgeons and technical staff should avoid excessive leaving and entering the surgical suite.

7.4.6. Scrub brushes for washing hands must be completely immersed in a clean container of an appropriate disinfectant (Appendix 15: Disinfectants).

7.4.7. The hand-brush container should be cleaned and re-filled with solution daily, and the brushes replaced daily with a new disposable brush or autoclaved brush. The scrub brushes must also be replaced if they are worn out. Brushes and disinfectant must be replaced in the course of the day if there is contamination or residual debris that may prevent effective disinfection.

7.4.8. Immediately prior to putting on sterile surgical gloves, the surgeon must do a standard surgical hand and forearm scrub with appropriate disinfectant (Appendix 15: Disinfectants) for 5-7 minutes. The hands are then rinsed with clean (preferably free–flowing, treated tap) water. They are then air-dried or dried with a sterile towel before a fresh pair of sterile surgical gloves are put on.

7.4.9. Hand scrubs following the first one of the day:

- If the surgeon performs an uninterrupted series of surgeries, without handling animals between surgeries, the full 5-minute hand scrub between patients may not be necessary. The used surgical gloves are removed, the hands are washed with disinfectant soap or with disinfectant gel (e.g., benzalkonium/alcohol gel), and the next pair of sterile surgical gloves is put on.
- If the surgeon handles animals between surgeries while wearing examination gloves, hands are scrubbed for a minimum of 1 minute with disinfectant, and then cleaned with disinfectant gel before a fresh pair of sterile surgical gloves is put on for the next surgery.
- If the surgeon handles animals without examination gloves, a full 5-7 minute hand scrub must be done before each surgery.
- It is preferable to wash the forearms, rather than to scrub them. Micro-abrasions in the surgeon’s skin may pose more risk of contamination than less forceful scrubbing does.

7.4.10. Hands may be air-dried or dried with a sterile towel. Do not dry them with non-sterile material. Once the surgeon's hands are disinfected for surgery, they must be kept in front of the body and must not touch a non-sterile surface. If there is any breach of sterility, the surgeon must scrub again.

7.5 Surgical gloves and drapes

7.5.1. New, sterile, correctly-sized, disposable surgical gloves that are within the factory-specified expiration date must be worn for each surgical patient.

7.5.2. Gloves that have been stored in extreme temperatures may be friable and not appropriate to use even if they are still within the expiration date.

7.5.3. Fresh, sterile drapes (disposable or autoclaved) must be used for each patient.

7.5.4. Dry, sterile drapes must completely cover the body of the patient for the duration of the surgery. The draped surgical field must be large enough to ensure adequate sterile space for the surgeon to work comfortably without risk of breaking sterility.

7.5.5. Male cats undergoing routine orchiectomy (castration) need not be draped.

7.5.6. Sterile drapes must be handled only by the surgeon after she or he has scrubbed hands and put on sterile gloves. The animal's skin must be cleaned and disinfected as outlined above before the surgical drape is placed.

7.5.7. The fenestration in the drape must allow visualization of the entire length of the incision site, but no more. Fur from the clip margin must not be visible inside the fenestration.

7.5.8. All reasonable precautions must be taken to prevent the drapes from getting wet before or during surgery.

7.6 Surgical instruments

7.6.1. A fresh, autoclaved surgery pack must be used for each patient. The single exception to this is for castration of male cats, in which case chemically-sterilized instruments may be used (cf. Appendix 15: Disinfectants).

7.6.2. Some projects use chemically-sterilized instruments. All efforts must be made to advance to the use of autoclaved instruments as a priority in the project's development. Chemical sterilization may be used in the interim, provided that the method has been tested in a controlled clinical setting with good patient follow-up, and that there are no post-operative complications that can be ascribed to failure of sterile technique or to adhesions caused by the chemicals.

7.6.3. A complete set of sterile, surgical instruments must be included in each pack. (cf. Appendix 2: Basic instrument pack for spay and neuter).

7.6.4. Instruments must be kept in good condition by ensuring that they are used and cleaned properly. They should be replaced when they cannot be used effectively and safely.

7.6.5. After use in surgery, all surgical instruments must be cleaned under cold water with dish soap or instrument cleaning solution to remove all visible organic matter. Particular attention should be paid to the serrated sections and hinges of the instruments to ensure that all organic matter has been removed. A toothbrush works well for this purpose.

7.6.6. Instruments are dried and, ideally, sprayed with a surgical instrument lubricant to extend the longevity of moving parts. Instrument packs are assembled in clean cloths, per Appendix 2: Basic instrument pack for spay and neuter.

7.6.7. Autoclave indicator tape or a stop tube must be used to ascertain sterilization.

7.6.8. The pack must be labeled with the date on which it was autoclaved. Autoclaved surgical packs must be used within 2 weeks of the autoclave date. Instruments or packs wrapped in plastic autoclave sleeves may be stored longer, per recommendations of the manufacturer of the plastic sleeves.

7.6.9. Autoclaved instrument packs must be stored in a cupboard or closed container in which they are dry and protected from dust and other contaminants.

7.6.10. If there is any question that the packs may have become contaminated or remained moist inside, they must be re-sterilized prior to use in surgery.

7.6.11. Autoclave machines should be maintained and serviced regularly, to ensure that they are in good working order.

7.6.12. Any surgical instrument, swab, blade, suture material or other sterile object that is contaminated during surgery must be discarded and not used again unless it can be effectively sterilized. It is not acceptable simply to rinse the item with alcohol or disinfectant and re-use it.

7.6.13. A fresh, sterile blade must be used for each surgical patient.

7.6.14. All suture materials must be sterile and handled in a way to maintain sterility. Foil packs must be opened outward so their content and inner surface remains sterile. The protruding end of suture from a reel or cassette should be pulled upward to expose only sterile suture for the surgeon to cut and handle.

7.6.15. Sterile technique is imperative for procedures that enter the peritoneal cavity and for castration of dogs. Any break in sterility, no matter how small, must be immediately corrected before surgery may proceed.

7.7 Technique for surgical sterilization of dogs and cats

7.7.1. Surgery should be performed in accordance with surgical techniques described in Small Animal Surgery (T.W. Fossum, Mosby, 1997). If there is any concern that surgical techniques do not meet acceptable standards, advice should be sought from the CA Project Manager or the IFAW veterinarian in charge of the project.

7.7.2. Each surgeon must be assisted by a person with good animal handling skills and capable of basic veterinary nursing skills to reliably perform such tasks as monitoring anesthesia, drawing up and injecting drugs, maintaining sterility, monitoring patient recovery, and assisting with surgery if necessary.

7.7.3. All surgical procedures must be done aseptically. Any break in sterility during the procedure must result in immediate measures to rectify asepsis. Subsequent measures must be taken to limit the risk of post-surgical infection or other complications.

7.7.4. Midline vs. flank approach for ovariohysterectomy

- Ovariohysterectomy for bitches should be via a midline approach. A flank approach may be used for small, non-pregnant bitches at the discretion of the surgeon. Some surgeons have found reliable outcomes with a flank approach even in larger dogs but it must be noted that surgical experience and good post-operative observations are essential to this technique.
- Queens may be spayed via a midline or flank approach.
- All pregnant cats and dogs, or animals with reproductive abnormalities (e.g., pyometra) must be spayed via a midline approach.

7.7.5. Bilateral orchiectomy for males should be via a scrotal approach in cats. Dogs are usually castrated by a pre-scrotal approach. Depending on the surgeon's experience and skill, a scrotal (perineal) approach may be used as well. The latter is advised for EAS (Section 8.6: Special surgical considerations for early-age sterilization).

7.7.6. Cryptorchid testes must be removed in all cats and dogs.

- The position of the retained testicle may be anywhere from inguinal, just cranial to the scrotum (where it is palpable beneath the subcutaneous tissue in the groin), to just caudal to the kidney.
- Removal of testes located in the abdominal cavity requires a standard, mid-line laparotomy. Surgical preparation of the abdomen must be done as for females.

7.7.7. All fetuses older than 3-4 weeks of gestation must be euthanized by injection with pentobarbital sodium intraperitoneally. If pentobarbital is not available, an overdose of another anesthetic may be used. Fetuses at this age are believed to be sentient and must not be left to die by asphyxiation from loss of placental oxygen.

7.8 Sutures

7.8.1. Suture material

- Suture material be sterile, of biomedical grade, approved for medical use, and dated for current use. Suture material must be of adequate tensile strength to ensure complete knot security and hemostasis.
- Some practitioners have found the use of chromic surgical gut ('catgut') suitable, but this must be used only with the recognition that catgut has a relatively rapid break-down time. Animals whose abdominal wall has been closed with catgut should be monitored with particular attention until they can be sure to have healed (7-10 days).
- Sutures that are left in the body (e.g., for ovarian pedicles, uterine stump, blood vessels, spermatic cords, muscle or under the skin) must be absorbable material or inert, non-absorbable material, such as stainless steel, nylon or polypropylene.

7.8.2 Closure of the abdominal wall

- The abdomen of dogs must be closed in 3 layers (body wall at the linea alba, subcutaneous layer, and skin sutures (intradermal (subcuticular) or skin).
- Cats: midline incisions may be closed in 2 layers (body wall at the linea alba, and skin) if there is little subcutaneous tissue. If there is more subcutaneous tissue, or if skin was undermined, it should be closed in 3 layers as for dogs. Flank incisions must be closed in 3 layers (each of the two abdominal muscle layers individually, and skin).
- The abdominal wall may be closed with a simple interrupted or continuous suture pattern placed in the linea alba (midline approach) or in the two separate abdominal muscle layers (flank spay). The simple interrupted pattern is highly recommended due to increased security.
- In bitches or queens for whom adequate post-operative observation of the surgical site is questionable, a simple interrupted suture pattern must be used to close the abdominal wall in order to minimize danger of wound dehiscence.

7.8.3 Skin sutures

- The skin may be closed with an intradermal (subcuticular) suture pattern or with skin sutures. Absorbable sutures are ideally placed intradermally.
- Skin adhesive may be applied to the outer surface of a well-opposed surgical skin wound. Do not use surgical glue within the wound, as it may contribute to granuloma formation.
- Non-absorbable skin sutures must be removed from dogs and cats once the wound has healed and appears normal (usually 10-14 days post-surgery). If it is unlikely that the animal will be returned for suture removal, or if the animal is too fractious to remove sutures safely, then intradermal sutures, preferably with an absorbable suture material, must be used.

7.9 Identification of sterilized animals

7.9.1. Animals who roam free or who do not have an owner should be permanently marked as sterilized, so that they are not unnecessarily caught and subjected to surgery again.

7.9.2. Ear notching of dogs

- This procedure should be done aseptically under general anesthesia at the time of sterilization.
- A 1.5 cm deep x 1 cm wide 'V' shaped notch along the distal half of the cranial margin of the left pinna should be removed. The size of the notch to be removed should be adjusted according to the size of the ear. Two hemostats (artery forceps) can be placed in a V-shape before the notch is cut with scissors.
- Preferably, the ear should be notched at the beginning of surgery, and the hemostats left in place until the end of surgery. This usually allows sufficient time for hemostasis without having to prolong anesthesia.
- Hemostasis may also be achieved by surgically cauterizing the cut skin margins.

7.9.3 Tattoos

- In bitches with long hair on their ears (where the fur would obscure the notch), or bitches amenable to handling so that their medial pinnae can be examined, tattooing may be used instead or in addition to ear notching. It should be kept in mind that tattoos fade with time.
- The surgical incision site may be tattooed for identification of spayed females.

7.9.4 Ear tipping of cats

- This procedure should be done aseptically during general anesthesia (at the time of sterilization).
- A hemostat (artery forceps) is clamped across the distal centimeter (as much as the distal quarter) of the left ear pinna, and the ear tip is removed by cutting straight across with scissors or a blade.
- Preferably, the ear should be tipped at the beginning of surgery, and the hemostat left in place until the end of surgery. This usually allows sufficient time for hemostasis without having to prolong anesthesia.
- Hemostasis may also be achieved by surgically cauterizing the cut skin margins.

7.10 Post-surgical care and recovery

7.10.1. A bandage may be placed over the surgical site if the cleanliness of the bandage can be ensured. Bandages must be changed at least every 12 hours, or more frequently if they become soiled. If bandages cannot be changed when they are soiled, it is better to not use any.

7.10.2. Elizabethan collars may be necessary for some owned animals to protect the surgical wounds. The aftercare of these animals should be carefully monitored, and their guardians shown how to remove and replace the collar as necessary. If a high proportion of animals are requiring a collar post-surgically, then the method used to clip the fur, surgical technique and wound closure must be re-assessed.

7.10.3. Post-operative antimicrobials

- Many veterinarians for field sterilization projects are in the habit of administering one-time injections of an antibiotic (usually a penicillin) at the time of surgery. It should be noted that this practice is not supported by the Association for Shelter Veterinarians spay/neuter guidelines, as it contradicts responsible use practices of antimicrobials.
- Routine use of antimicrobials must never be substituted for aseptic technique and excellent sanitation practices.
- Antimicrobial use should be reserved for specific indications, such as preexisting infection (e.g., pyometra), in which case therapy should be started prior to surgery when at all possible with an appropriate choice of antimicrobial. Therapy must continue post-operatively at the appropriate dose for the appropriate duration of time.
- Where a break in surgical sterility is recognized, or if the patient develops infection post-operatively, the appropriate choice of antimicrobial must be administered at the appropriate dose for the appropriate duration of time.

7.10.4. Drinking water and a small portion of food should be offered once the animal is able to walk and there is no risk that he or she will accidentally inhale water or drown in a water bowl.

7.10.5. Young animals (less than 16 weeks old) should receive food and water as soon as possible to avoid hypoglycemia. Food should be soft (e.g., tinned food), easy to consume and easily digestible. These patients may require encouragement with oral or intravenous glucose if they appear weak and unwilling to eat right away. Hypoglycemia is an important concern in these patients. Refer to Section 8.2: Pediatric physiology in anesthesia.

7.10.6. It is essential to keep patients warm during recovery. This is particularly true for pediatric patients. During anesthesia, thermoregulation is compromised, and young animals may quickly become hypothermic.

7.10.7. An animal may be discharged after surgery once s/he can walk unassisted, is alert and vital sign parameters are normal.

7.10.8. Efforts should be made to monitor dogs and cats who won't be supervised in a clinic or by a guardian after surgery (e.g., in a trap-neuter-release project) for at least a few hours, if not overnight, after they have recovered from anesthesia. Ideally, they should eat and drink before release. These patients must demonstrate full cognitive and muscle control before release.

7.10.9. The guardian should be given detailed, written or illustrated instructions that explain post-operative care for the animal, and what problems to report to the veterinarian (Appendix 14: Examples of clinical forms and record sheets). Ideally, instructions are also explained verbally before the animal is taken home.

7.10.10. The patients' guardians must be given the name and telephone number of a veterinarian or clinic to contact, should the patient develop complications or if the guardian has questions.

7.10.11. All instances of infection, inflammation and other painful or untoward reactions or complications in patients post-surgery must be monitored and treated until they have resolved. The surgical team should address these reactions and complications and undertake steps to reduce their incidence to a minimum.

7.10.12. In a spay/neuter campaign, it is highly valuable to visit each patient, or to call the guardian, approximately one week after surgery to check on the patient's recovery, assess animal guardianship, answer owners' questions and to strengthen the bond between community members and project teams.

These post-operative visits may be perceived as unnecessarily time-consuming. However, it promotes a strong message of animal guardianship and the commitment of the veterinary team and of the entire project to the community. It also provides an invaluable opportunity to observe animal guardianship issues that need to be addressed in future and to discuss animal care with people who may otherwise not come forth to ask questions.

7.10.13. Post-operative visits may be made by a trained nurse and volunteers if a veterinarian is unavailable, but with a veterinarian available to the nurse/volunteer by telephone. Ideally, the nurse will have the ability to take photographs of wounds or other lesions to send to the off-site veterinarian (e.g., with a mobile telephone). This can greatly facilitate decisions and instructions. A supply of oral antibiotics, wound cleaning supplies, oral analgesic and parasite control medications should be taken along to treat those patients who can be managed *in situ* by their guardians. Patients with more serious complications or who do not have a guardian who can care for them adequately should be taken to a clinic.

7.10.14. In some projects, roaming dogs of uncertain ownership may be sterilized and released to roam again after they recover from anesthesia. Efforts should be made to engage the community in monitoring these animals post-operatively so that if someone observes an animal to be ill, behaving abnormally or exhibiting a wound, they should call the project leader or veterinarian to notify him or her of the problem. In areas where people have no access to veterinary service, consideration must be given to the availability of basic veterinary supplies (e.g., oral antibiotics, wound cleaning supplies, oral analgesic and parasite control medications) in the event that a patient needs post-operative support. Enlisting the community in this way helps to engage people in the care of roaming animals and a reason to recognize the existence of these animals in a compassionate context. Community members may enjoy the feeling of empowerment to help them. This may be a particularly powerful message for children, who are often highly observant of animals but are traditionally unheard and powerless members of the community.

8. Early-Age Sterilization (EAS)

8.1 Clinical considerations for early-age sterilization

8.1.1. EAS refers to puppies and kittens 6-16 weeks of age (minimum weight 800g)

8.1.2. EAS may be performed so long as the surgical team is familiar with, and prepared for, the special needs of pediatric patients.

8.1.3. All requirements must be met as for older dogs and cats as outlined in the sections above. In addition, the following physiological considerations for young animals must be taken into account.

8.1.4. As with adult dogs and cats, puppies and kittens must receive a full physical examination prior to surgery to ensure that they are in sufficiently good health to undergo surgery. If the patient is underweight, ill, heavily parasitized or otherwise debilitated, the health conditions must be treated and surgery delayed until s/he is in sufficiently good health for surgery to pose no more than a reasonable risk.

8.2 Pediatric physiology in anesthesia

8.2.1. Hypoglycemia

Young animals can become hypoglycemic quickly. This is due to a high metabolic rate and reduced hepatic glycogen stores compared with adult animals. Therefore:

- Anesthesia time must be kept to a minimum
- Small portions of food may be fed up to 30-60 minutes before anesthesia.
- If anesthesia lasts for more than one hour, IV fluids should contain 2.5-5% glucose or dextrose.
- Provide a small meal of easily-digestible food as soon as the puppy or kitten is sufficiently awake to eat safely. If the patient does not eat right away, give 3-5 ml oral 5% glucose or corn syrup to improve the animal's energy and appetite and to pre-empt hypoglycemia.
- Water must be available to animals at all times before anesthesia. Following recovery from anesthesia, water must be available again as soon as the patient is able to drink.
- Keep animals warm throughout anesthesia to prevent excessive energy loss to thermoregulation.

8.2.2. Hypothermia

- Pediatric patients become hypothermic easily due to their large body surface area-to-volume ratio, reduced ability to shiver, and little subcutaneous fat. Anesthesia further compromises thermoregulation.
- Hypothermia contributes to bradycardia, hypotension, and retarded metabolism of drugs. Increased energy use to maintain euthermia increases the risk for hypoglycemia.
- Anesthesia time and surgery time must be kept to a minimum.
- Patients must be insulated from the surface on which they are lying (thick towel, blanket, thick cardboard).
- Patients must be kept dry. Avoid the use of alcohol on skin. Use a non-alcohol skin prep to prepare the surgical site (e.g., chlorhexidine).
- Supplemental heat must be provided to pediatric patients throughout anesthesia.
- Supplemental heat sources may be provided in the form of electric heat pads, microwavable liquid heat pads, warm water bottles, or socks filled with (uncooked) rice, barley, lentils or other small grains or legumes and heated in the microwave.
- Care must be taken that the heat source is not so hot that it will scorch the skin. Heat sources should be approximately 38°C. Check the temperature as you would a baby's bottle: hold it against your inner wrist. It should feel pleasantly warm. If you feel the need to remove your hand within 60 seconds, then it is too hot. If it feels cooler than your skin, then it is too cold.

- A towel or blanket or thick cardboard must always be placed between the heat source and the patient's skin.
- Rectal temperature must be monitored each time that vital signs are assessed (every 5 minutes). If the puppy or kitten begins to shiver, paws and ears feel cold, or rectal temperature falls below 37°C, additional heat sources, blankets and warmed IV fluids may be necessary to raise body temperature. Steps to warm the patient must be taken immediately. Animals must be supervised until they are stabilized.

8.2.3. Hepatic function

- Hepatic function matures in puppies and kittens at 12-14 weeks of age. Younger animals have low glycogen stores, which predisposes pediatric patients to hypoglycemia. The measures outlined above to prevent hypoglycemia are critical for pediatric patients.
- Anesthetic drugs that are metabolized by the liver will be excreted more slowly.
- Plasma albumin levels are lower than in adults, so pediatric patients are more sensitive to protein-bound drugs. Conversely, pediatric patients initially will appear less responsive to drugs that have little protein binding.

8.2.4. Respiratory system

- Pediatric patients have smaller airway diameters and more flexible airway cartilage. The risk of airway obstruction is therefore greater than in adults, and all patients must be intubated.
- Care must be taken to prevent airway trauma during intubation, per instructions in Section 6.3: Endotracheal intubation. Endotracheal tubes must be not too big or small, cuffs must be inflated and deflated with care, and the tubes must be lubricated appropriately prior to placement.
- Kittens up to ca. 1.5 kg will take a 3.0 mm endotracheal tube. For larger kittens, try 3.5 mm. Puppies will usually take 3.5 – 4.5 mm tubes.
- Kittens must always receive topical lidocaine prior to placement of the endotracheal tube, as described in Section 6.3: Endotracheal intubation.
- The metabolic requirements of pediatric patients are 2-3 times higher than for adults. The functional residual capacity in the airways and smaller tidal volumes result in smaller oxygen reserves and a faster respiratory rate than in adults. Therefore:

1) Pediatric patients will need oxygen supplementation throughout anesthesia if they are not being maintained on inhalant anesthetic.

2) Respiratory rates should be 2-3 times those of adults dogs and cats.

3) Induction with gas anesthetics is more rapid than with adults

4) Avoid the use of respiratory depressant drugs (e.g., alpha-2 agonists such as xylazine and dex/medetomidine)

5) Gentle, positive-pressure ventilation (PPV) should be provided intermittently (ca. every 2-3 minutes) throughout anesthesia, particularly to those patients with low or shallow respiratory rates.

8.2.5. Cardiovascular function

- A greater portion of the cardiac output in pediatric patients goes to the brain than in adults. Therefore, young animals are more sensitive to intravenous and inhalant anesthetics.
- In very young animals, the ability of the heart to increase cardiac output and the vasoconstriction of blood vessels are less than in adults. Therefore, the physiologic compensation to low blood pressure and the response to fluid therapy to maintain blood pressure will be reduced in comparison to adult animals. Therefore:
  - It is critical that the heart rate in young patients remains above 150 bpm (normal ca. 200) during anesthesia and surgery.
  - Shock, particularly hypovolemic shock (e.g., due to hemorrhage) may be very difficult to reverse.
  - IV fluid support should be given if surgical procedures last longer than 30 minutes. The rate should be carefully controlled at 10 ml/kg/hour, and respiration monitored closely for evidence of pulmonary edema. Fluids given too quickly may drown the patient. Fluids given too slowly may result in dehydration.
  - Normal crysalloids (physiologic saline (0.9% NaCl), Hartmann's or Lactated Ringer's solution) are appropriate for normal circumstances. If hypotension is a concern, colloids (Hetastarch) is valuable (5-10 ml/kg/hr).

8.2.6 Renal function

- Renal function matures after 8 weeks of age.
- Care must be taken to prevent over-hydration when delivering intravenous fluids (risk of pulmonary edema). Care must also be taken to prevent dehydration of the patient.
- During surgery, IV fluids must be run at 10 ml/kg/hour, and no faster.
- Monitor respiration closely and beware of the development of pulmonary edema.
- Sterile 0.9% NaCl (saline) or Lactated Ringer's solution may be given subcutaneously, maximum 10 ml per injection site.
- Do not give glucose or dextrose subcutaneously. Glucose must be given only orally or intravenously.

8.3. Anesthesia for early-age sterilization: general principles

8.3.1. Stress must be avoided prior to anesthesia and throughout recovery.

- Puppies and kittens must be kept with littermates or in the environment in which they feel most comfortable until they are anaesthetized.
- Keep animals warm and comfortable, and handle them very gently.
- Avoid loud noises, excessive activity, isolation, distressed animals nearby, intimidation by other animals, and other sources of stress.
- Pre-anesthetic stress compromises the animal's response to anesthetic drugs.
- Stress predisposes pediatric patients to hypoglycemia.

8.3.2. Duration of anesthesia must be kept to an absolute minimum. All drugs, supplies and equipment necessary for anesthesia and surgery must be prepared fully before the animal is anesthetized.

8.3.3. All considerations for anesthesia as outlined in Section 6: Anesthesia must be followed. Each puppy or kitten must be intubated, and must have an IV catheter in place.

8.3.4. Pediatric patients should receive supplemental oxygen during anesthesia even if not receiving a gas anesthetic. If this is not possible, gentle, positive-pressure ventilation must be supplied every 2-3 minutes to ensure sufficient ventilation.

8.3.5. Anesthesia must be monitored as described in Section 6.6: Anesthetic monitoring.

8.4 Anesthetic premedication for early-age sterilization

8.4.1. An anticholinergic drug such as atropine and glycopyrrolate should always be administered as premedication to maintain heart rate for pediatric patients (cf. Section 8.2: Pediatric physiology in anesthesia)

8.4.2. Phenothiazine tranquilizers (acepromazine) should be avoided in pediatric patients, as they may produce prolonged CNS depression and potentiation of hypotension and hypothermia.

8.4.3. Benzodiazepines (diazepam, midazolam) result in minimal sedation when used alone. However, when added to an opiate they may have a synergistic sedative effect and, when added to ketamine, will improve muscle relaxation.

8.4.4. Alpha-2 adrenergic agents (xylazine, medetomidine) slow the heart rate and decrease contractility. Given the cardiovascular concerns in pediatric patients (Section 8.2: Pediatric physiology in anesthesia), these drugs should be avoided. If they are used, animals must be pre-medicated with an anticholinergic and vital signs monitored very closely throughout anesthesia and recovery.

8.4.5. Opioids cause sinus bradycardia and respiratory depression. An anticholinergic should be given as premedication. Adult dose rates of opioids should be halved for pediatric patients. Vital signs must be monitored very closely throughout anesthesia and recovery.

8.5 General anesthesia for early-age sterilization

8.5.1. Propofol is the induction agent of choice for pediatric patients.

- Patients should be pre-oxygenated prior to receiving propofol.
- Pediatric patients should not receive repeated doses of propofol, as it is metabolized by the liver and repeated doses may prolong recovery.

8.5.2. Isoflurane may be used for induction (mask).

8.5.3. Isoflurane gas anesthesia is the method of choice for anesthetic maintenance in pediatric patients. Isoflurane is minimally processed by the liver, and allows rapid adjustment of anesthetic depth and rapid recovery.

8.5.4. Barbiturates (pentobarbital, thiopentone) depend entirely on hepatic metabolism for termination of effects and are therefore contraindicated in puppies and kittens less than 12 weeks old.

8.5.5. Recovery from diazepam/ketamine and zolazepam/tiletamine depends on hepatic metabolism and renal perfusion. Care must be taken with the use of these drugs in animals younger than 12 weeks of age.

8.5.6. Refer to Appendix 5: Anesthetic and analgesic drug protocols for further discussion of anesthetic drugs.

8.6 Special surgical considerations for early-age sterilization

8.6.1. Puppies normally have more peritoneal fluid than adult dogs. The surgeon must anticipate this.

8.6.2. Tissues are more friable and delicate than those of adults. Gentle tissue handling and surgical experience are imperative.

8.6.3. The abdominal incision in female puppies is made further caudally than in the adult female, as the ovarian suspensory ligaments are not as tight.

8.6.4. Meticulous hemostasis is essential, as even minimal hemorrhage can be significant in a pediatric patient.

8.6.5. Closure of the abdomen is the same as in adults, although skin sutures should be avoided, as puppies and kittens tend to chew on them.

8.6.6. It is recommended to avoid the use of polydioxanone (PDS) in subcutaneous and intradermal layers to avoid the development of calcinosis circumscripta. Polyglactin (e.g., Vicryl®) is a good choice.

8.6.7. Male puppies may be castrated by a standard pre-scrotal approach like adult dogs, or through a scrotal approach like male cats.

8.6.8. For the scrotal approach:

- The surgical site must be prepared for a sterile procedure (unlike in cats, in which it is a clean procedure.)
- A single incision can be made to remove both testicles.
- The cord may be tied on itself or ligated with absorbable suture material. Incisions are not sutured, and are left to heal by second intention as in male cats.

8.6.9. Male kittens are castrated in the same manner as adult toms (one incision per scrotum, clean (not sterile) procedure). The cord may be tied on itself or ligated with suture material.

8.6.10. If both testicles are not palpable in the scrotum, castration should be delayed until 6 months of age. At that age, both testicles should be descended; if not, the retained testicle must be retrieved from the inguinal or abdominal area. Retained testicles must be removed.

8.7 Post-surgical care

8.7.1. Provide a small meal of easily-digestible food as soon as the puppy or kitten is sufficiently awake to eat safely. If the patient does not eat right away, give 3-5 ml oral 5% glucose or corn syrup to improve the patient's energy and appetite and to avoid hypoglycemia.

8.7.2. Provide supplemental warmth until patients are fully recovered and have eaten.

8.7.3. For 5-7 days after surgery, puppies and kittens should be kept in a cage that restricts excessive activity.

8.7.4. Puppies and kittens who are housed together after sterilization surgery must be observed closely to ensure that they don't compromise their own or one another's surgical wounds.

9. Vaccination and parasite control

9.1. While vaccination and parasite control are not within the immediate protocol of sterilization surgery, presentation of the animals for sterilization surgery provides an excellent opportunity to provide these basic services, particularly for dogs and cats who may be otherwise unlikely to receive them.

9.2. Dogs and cats can be sterilized, vaccinated and dewormed all in the same day. Ideally, animals are vaccinated and dewormed ca. 2 weeks prior to surgery. But if this is not feasible, all three can and should be done at once.

9.3. All dogs and cats 3 months of age or older and living in rabies-endemic areas must be vaccinated against rabies according to international guidelines or local legislative requirements (Appendix 3: Vaccination Guidelines).

9.4. All dogs and cats should be vaccinated with core vaccines according to recommended schedules outlined in Appendix 3: Vaccination Guidelines.

9.5. Project resources may restrict the ability to fully vaccinate animals according to guidelines with core vaccines other than rabies. This will result in otherwise avoidable disease and mortality, but it is a realistic and unfortunately common situation that must be acknowledged under practical field guidelines. Under these conditions:

9.5.1. Rabies vaccination for all dogs and cats older than 3 months of age must be considered essential and non-negotiable.

9.5.2. Animals less than 1 year old should be vaccinated at least once, immediately on rescue or removal from the dam. As many boosters as possible should be given thereafter according to guidelines (Appendix 3: Vaccination Guidelines), particularly for puppies and kittens younger than 6 months of age.

9.5.3. If possible, adults should be vaccinated at least once on arrival as well.

9.5.4. Disease outbreak must be expected in the face of substandard vaccination.

9.5.5. Biosecurity measures are always important, and are particularly critical in situations where animals are inadequately vaccinated. Any sign of illness in animals must be immediately addressed with isolation, treatment or removal, disinfection, and control of fomite movement.

9.6 Biosecurity measures for infectious disease control may be found in shelter medicine texts such as that of Miller & Zawistowski, 2004. Basic principles include:

9.6.1. Prevent exposure of susceptible animals to animals who are ill or who may be infectious. Exposure may occur through direct contact among animals, through contact with excretions and secretions from animals who are shedding pathogens, transfer of pathogens on the hands of people handling animals, or on fomites.

9.6.2. Follow strict quarantine and isolation procedures for sick and potentially infectious animals.

9.6.3. Follow strict personal hygiene protocols to prevent transmission of infectious pathogens to susceptible animals on hands, clothing, shoes and other fomites.

9.6.4. Follow strict cleaning and disinfection protocols to minimize contamination of animal areas.

9.6.5. Ensure that animals are well fed, warm, dry, clean and comfortable and are otherwise as much as possible free of stress and boredom. Stress plays a strong role in immunocompromise and disease susceptibility.

9.7 Modified live vaccines should be avoided for clinically ill, debilitated or pregnant animals. However, when large groups of animals are rescued all at once, or in shelter medicine protocols, it is strongly advised to vaccinate every animal immediately. These animals must continue to be managed as vulnerable to contagious disease, as their immune response to the vaccination may be sub-optimal. Although the antibody titers in compromised animals may not be as strong as in a healthy animal, vaccination in these cases will still afford a certain level of immunity and will help to mitigate the extent and severity of disease in the group or shelter.

9.8 Protocols for the control of endoparasites must address prophylaxis against parasites endemic in the area. Refer to Appendix 4: Anti-parasitic drugs for drugs and usage guidelines.

9.9 Protocols for the control of ectoparasites are based on the needs of individual patients. Refer to Appendix 4: Anti-parasitic drugs for drugs and usage guidelines.

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10. Euthanasia

10.1. Refer to the document "The welfare basis for euthanasia of dogs and cats and policy development" prepared by the International Companion Animal Management Coalition (ICAM). This document provides structured, practical guidance for developing a euthanasia policy, how to decide whether euthanasia is a reasonable option for an individual animal, and for guidelines on how to perform euthanasia. The document may be found at www.icam-coalition.org under "Tools and Resources".

10.2. Euthanasia may be a consideration for the conditions of some patients. The decision must be made on the basis of a rational consideration of options available to the animal and to the people responsible for the animal's care. The decision to euthanize an animal may be made for any of five reasons:

10.2.1. Medical

- An animal who is suffering from an acute or chronic disease, condition or pain that cannot be alleviated to a satisfactory degree, given the practical and financial resources available. Suffering can be defined here as a restriction of any or all of the five welfare needs.
- An animal who is suffering from an acute or chronic disease or illness that might pose a risk to other animals or to people, particularly if appropriate preventative measures are not in place (e.g., rabies).

10.2.2. Behavioral

- An animal with a behavioral problem that results in suffering due to the animal experiencing fear and distress, and that cannot be successfully treated with behavior therapy considering the constraints on practical and financial resources available.
- An animal with a behavioral problem that presents a risk to him or herself, to other animals, to people or to the environment and that cannot be successfully treated considering the constraints on practical and financial resources available.
- An animal who cannot be re-homed because of a behavioral problem that cannot be corrected considering constraints on practical and financial resources.

10.2.3. Lack of resources

- An animal who cannot be looked after or treated due to lack of finances, staff, expertise, suitable equipment or facilities and who will suffer as a result.
- An animal who is holding space in a shelter over a long period (e.g. because he or she cannot be re-homed) that could be used to benefit a large number of other animals.

10.2.4. Inadequate guardianship

- An animal whose needs (as identified by the five welfare needs) cannot be met due to a lack of owner or adequate guardian.

10.2.5. Legal order

- An animal who has been ordered by law to be euthanized, e.g. for disease control.

Appendices

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Appendix 1: Normal clinical parameters for dogs & cats

From the Veterinary Merck Manual, 10th edition 2010 and *Veterinary Pediatrics* by J.D. Hoskins, 3rd ed. 2001.

	ADULT DOGS	ADULT CATS
Heart rate (beats per minute)[1]	70-120	120-140
Respiratory rate[2] (breaths per minute)	18-34	16-40
Temperature[3] (rectal) ºC	37.9 – 39.9	38.1 – 39.2
Urine specific gravity	1.016 – 1.060 (usually > 1.030)	1.020 – 1.040 (usually > 1.035)
Urine volume (ml/kg/day)	20-100	10-20
PCV (hematocrit), %[4]	35-57	30-45
Hemoglobin g/dL	12-19	10-15
Red blood cells ($x10^{12}$/L)	5.0-7.9	5-10
White blood cells ($x10^9$/L)	5.0-14.1	5.5-19.5
Neutrophils (%, $x10^9$/L)	58-85, 2.9-12.0	45-64, 2.5-12.5
Band neutrophils (%, $x10^9$/L)	0-3, 0-0.45	0-2, 0-0.3
Lymphocytes (%, $x10^9$/L)	8-21, 0.4-2.9	27-36, 1.5-7.0
Monocytes (%, $x10^9$/L)	2-10, 0.1-1.4	0-5, 0-0.9
Eosinophils (%, $x10^9$/L)	0-9, 0-1.3	0-4, 0-0.8
ALT (U/L)	10-109	25-97
Alkaline phosphatase (U/L)	1-114	0-45
AST (U/L)	13-15	7-38
GGT (U/L)	1-9.7	1.8-12
Creatine kinase (U/L)	52-368	69-214
LDH (U/L)	0-236	58-120
Bilirubin (mg/dL; mmol/L)	0-0.3, 0-5.1	0-0.1, 0-1.7
BUN (mg/dL, mmol/L)	8-28, 2.9-10	19-34, 6.8-12.1
Creatinine (mg/dL, mmol/L)	0.5-1.7, 44-150	0.9-2.2, 80-194
Blood glucose[5] (mg/dL, mmol/L)	76-119, 4.2-6.6	60-120, 3.3-6.7
Total protein (g/dL)	5.4-7.5	6.0-7.9
Albumin (g/dL)	2.3-3.1	2.8-3.9
Globulin (g/dL)	2.4-4.4	2.6-5.1
Ca (mg/dL, mmol/L)	9.1-11.7, 2.3-2.9	8.7-11.7, 2.2-2.9
P (mg/dL, mmol/L)	2.9-5.3, 0.9-1.7	3.0-6.1, 1.0-2.0

1. Heart rates in puppies and kittens is up to 200; in kittens up to 240.
2. Respiratory rate for puppies and kittens is 2-3x that of adults due to higher metabolic requirements and smaller tidal volume.
3. Body temperature in puppies & kittens younger than 4 weeks of age is lower than in adults, and an external heat source is essential.
4. PCV (hematocrit) in puppies and kittens is normally lower than in adult dogs, in the range of 25-34 %
5. In cats, hyperglycemia (10-12 mmol/L, 180-216 mg/dL) as a consequence of stress is not uncommon and is not necessarily physiologically abnormal.

Appendix 2: Basic instrument pack for spay and neuter surgery

Instrument	Number of each
Backhaus towel clamps	4
#3 Scalpel handle (not mandatory, but helpful)	1
#10 Scalpel blade	1
Adson-Brown Tissue forceps or 1 Adson (rat-tooth) tissue forceps + 1 atraumatic (smooth-ended) dressing forceps	1 or 2
Halsted mosquito hemostats (for pinpoint bleeding and small vessels)	2
Kelly or Dunhill hemostats (or similar medium sized hemostat for pedicle crushing and ligation)	4
Scissors (blunt-tipped)	1
Mayo-Hegar or Olsen-Hegar needle holder	1
Ovariohysterectomy hook (optional but very helpful)	1
Needles, assorted sizes	2 or 3
Gauze sponges (10 x 10 cm)	10
Cloth surgical drape, fenestrated, large enough to cover the animal*	1
Surgical towels (additional draping or drying hands)	1 or 2
Sterile, gauze sponges in separate package in case more are needed	25/pack

* The cloth surgical drape may be omitted if disposable surgical drapes are used.

Instruments must be wrapped in a clean drape to create the instrument pack. That pack must be wrapped in a second drape and sealed with autoclave indicator tape. Alternatively, instruments may be sealed in autoclave bags or packed in a sealed instrument tray.

Autoclave conditions necessary for sterilization are 134°C for 3 minutes or 121°C for 15 minutes at 15 psi (100kPa) above atmospheric pressure.

Packs must dry inside the autoclave before they are removed from the machine. If using a pressure-cooker style autoclave, place a clean brick in the bottom and the surgery packs on top of the brick. The brick will help to absorb moisture and prevent the pack from emerging from the pot dripping wet.

Items that are sterilized by autoclave must have autoclave indicator tape on the outside of the pack, marked with the date on which the pack was autoclaved. Ideally, an autoclave indicator strip is placed inside the pack as well, among the instruments, to verify that the inside of the pack reached sufficient conditions for sterilization.

Autoclaved instrument packs must be stored in a cupboard or closed container in which they are dry and protected from dust and other contaminants. They must be used within 2 weeks of the sterilization date. (Packs wrapped in sealed plastic autoclave sleeves may be stored longer, per recommendations of the manufacturer of the plastic sleeves.) If there is any question that the packs may have become contaminated or remained moist inside, they must be re-sterilized prior to use in surgery.

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Appendix 3: Vaccination Guidelines

Core vaccines are those recommended for all cats and dogs. These are parvo, distemper, adenovirus-2 and rabies for dogs; and herpesvirus-1, calicivirus, parvo (panleukopenia) virus and rabies for cats. Non-core vaccines are administered to animals who are in specific risk categories, either individually or because of the shelter or rescue environment. Evaluation of the need for non-core vaccines must be based on a risk/benefit analysis that includes considerations of individual animal health, herd health, laboratory data, and resource allocation. Vaccination must never be substituted for good biosecurity practices.

The core vaccines with which dogs and cats are immunized are usually live attenuated (modified live, MLV) virus or recombinant vaccines. The notable exception is the rabies vaccine, which must always be killed virus or recombinant vaccine. Other than rabies, killed/inactivated virus vaccines are generally reserved for animals who are immunocompromised (e.g., cats infected with FIV or FeLV) or pregnant.

Vaccination of pregnant or lactating dogs and cats is generally not recommended due to the risk that the MLV reverts to virulence and infects the fetuses. When at all possible, queens and bitches should be properly vaccinated before they become pregnant. However, if the animal first presents when pregnant or lactating and is unvaccinated, or the vaccination history is unknown, as in a rescue situation, the risk of disease exposure in the environment must be assessed and balanced against the need for disease control in the group. Alternatively, killed/inactivated or recombinant vaccines may be used, with awareness that resulting protection may be less than optimal.

As stated, MLV vaccines are not administered to clinically ill, debilitated or pregnant animals. However, when groups of potentially compromised animals of uncertain vaccination status are rescued, and in shelter medicine protocols, it is strongly advised to vaccinate every animal immediately on intake, with the exception of severely ill animals. The latter should be isolated and treated as a biosecurity risk: if an animal is so ill that he or she cannot mount an immune response to vaccine, the patient is most likely not suitable to be admitted into a shelter environment. The MLV core vaccines begin to protect the animal within just a few hours or days of vaccination, well before an antibody titer can be measured in the serum.

This rapid immunity can reduce the duration and degree of viral shedding (depending on the virus), and will reduce the severity and duration of disease outbreaks.

The role of vaccination in a rescue or shelter situation is not only to protect the individual animal against disease, but also to prevent the spread of pathogens and disease in the shelter or foster home environment. For this reason, it is imperative that dogs and cats be vaccinated immediately or even before arrival at the shelter. This is necessary even if animals are likely to be euthanized a few days or weeks later due to the shelter's policies on resource and space management. Animals sterilized in a trap-neuter-release (TNR) program should be similarly vaccinated with core vaccines.

If animals are ill or debilitated, they may not be able to mount a full immune response to vaccination. So long as these patients are afebrile and eating, they should still be vaccinated in rescue and shelter situations, but should be managed as "vulnerable" individuals in the shelter's biosecurity paradigm. Although the antibody titers may not be as strong as in a healthy animal, vaccination in compromised animals will still afford a certain level of protective immunity. Revaccination as soon as the animal is healthy (2-3 weeks later) is particularly important for these individuals. If re-vaccination is delayed for 6 weeks or more, the course of vaccination should be restarted.

Kittens and puppies are generally more vulnerable to infectious diseases than are adult cats and dogs. Animals younger than six months of age are a critical and primary population on which to concentrate vaccination efforts, particularly in a rescue or shelter situation.

A common misconception by local veterinarians in many project areas is that animals should not be vaccinated and dewormed at the same time, or at the time of sterilization. To the contrary: all three can be, and should be, done simultaneously. Ideally, animals are vaccinated at least 2 weeks before being taken to a clinic or sterilization event so that they are properly protected against circulating pathogens there, or so that they do not pose a risk to the other patients by shedding pathogen. But if this is not possible, then vaccination, deworming, ectoparasite treatment and sterilization should all be done at once.

Recent research by vaccine immunologists and shelter veterinarians has advanced the practical application of on-site antibody assays, particularly for the control of disease outbreaks (e.g., see publications of Dr. Ronald Schultz, Professor of Pathobiology, College of Veterinary Medicine, University of Wisconsin, Madison, USA and http://www.maddiesfund.org). The assays that have been validated by clinical virologists (VacciCheck™, Biogal and TiterCHEK®, Pfizer) are applicable for canine distemper, parvo and adeno viruses (core vaccine concerns). It is essential that assays are rigorously validated to correlate with "gold standard" assays and with laboratory vaccine-challenge studies. Assays with poor sensitivity (false negatives) or poor specificity (false positives) can be devastating if decisions are made on the basis of their results.

In a shelter or rescue situation, or in the face of a disease outbreak, these antibody assays may be used to quickly determine which individuals must be quarantined or – better yet – sent to a safe foster guardian until they have had time to mount a fully protective immune response to the vaccine (ca. 2 weeks). Strong and well-enforced biosecurity protocols and excellent record-keeping are essential to enable such a system to function. Accurate disease diagnosis is also essential to ensure that the assays are being used for the correct diseases. The main drawback of the on-site antibody assays are expense and the need for clinicians to be properly trained in how to interpret the assay results and how to apply them to a real-time disease outbreak or high-risk situation. It must be kept in mind that disease outbreaks in shelters or in rescues where animals are severely traumatized and debilitated often involve multiple pathogens (e.g., distemper, parvo and respiratory disease together).

Antibody assays are not to be used for decisions about whether or not to vaccinate animals in shelters, rescues or disease outbreaks, or against rabies. The immunology of many of these pathogens depends on local secretory immunoglobulins and non-humoral immunity that cannot be assessed with antibody assays. Animals should be vaccinated and handled according to protocols, and not on the basis of antibody titers.

Vaccines must be produced by a reputable manufacturer that adheres to internationally-recognized quality control protocols. The vaccines must be kept at the appropriate temperatures (4°C for most vaccines) until they are administered to the patient. Vaccines that become inappropriately cold or warm quickly become inactivated and no longer induce a reliable immune response in the patient. Lyophilized vaccines must be used immediately (within an hour) after reconstitution.

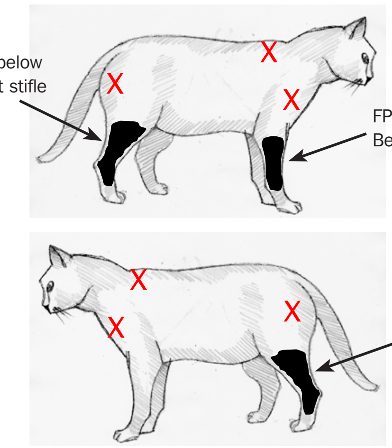
Good records for vaccination are important, both for the management of clinical services and to support the owner or guardian in responsible guardianship practices. Individual countries may require specific vaccination records by law, for example for rabies. Records are also important to document patterns of vaccine failures or vaccine reactions. It is advised to notify the vaccine manufacturer of adverse reactions if local laws don't preclude the necessity of this. Owners and guardians should be advised of clinical signs to report to the veterinarian in the 2-3 days following vaccination of their dog or cat. The risk of vaccination-site sarcomas in cats has prompted the development of vaccination location guidelines according to the diagram below. For further discussion of this issue, refer to the 2013 AAFP feline vaccination guidelines.

A dose of vaccine consists of the same volume for a puppy or kitten as for an adult dog or cat. Vaccine doses should not be halved or otherwise reduced for young animals or small individuals.

MLV = modified live virus **killed** = killed virus
parenteral = SQ or IM administration

SQ = subcutaneous **IM** = intramuscular *= core vaccines

Vaccines are usually given **SQ** (subcutaneously) unless specifically noted in the charts below. Some vaccines are designed to be administered **intranasally**. Intranasal vaccines must not be injected SQ or IM. Conversely, parenteral vaccines must never be injected intranasally or by any other route. The skin should not be disinfected prior to vaccination, as this may inactivate MLV antigens.



Recommended vaccination sites for cats

Canine vaccination guideline for general veterinary practice

(modified from AAHA 2011 canine vaccination guideline)

Canine vaccine	Initial puppy vaccine (< 16 weeks)[1]	Initial vaccination for dogs older than 16 weeks	Revaccination
*Canine Parvovirus (CPV2) MLV	Give one dose every 3-4 weeks between the ages of 6 and 16 weeks.	One dose is considered protective and acceptable.	• Puppies last vaccinated at 16 weeks of age should be revaccinated one year later, then once every 3 years. • Dogs first vaccinated when older than 4 months are revaccinated 3 years later, then every 3 years. • All commercially-available MLV-CPV2 vaccines currently protect against Canine Parvovirus 2a, 2b and 2c. • In healthy dogs, the protective immune response induced by the MLV-CPV2 vaccine is expected to last for at least 5 years. • Vaccine must be used within one hour of reconstitution, as it loses virulence rapidly. Ideally, it is administered to the patient immediately after reconstitution.

Canine vaccination guideline for general veterinary practice

(modified from AAHA 2011 canine vaccination guideline)

Canine vaccine	Initial puppy vaccine (< 16 weeks)[1]	Initial vaccination for dogs older than 16 weeks	Revaccination
*Canine Distemper Virus (CDV) MLV or rCDV[2]	Give one dose every 3-4 weeks between the ages of 6 and 16 weeks.	One dose is considered protective and acceptable.	• Puppies last vaccinated at 16 weeks of age should be revaccinated one year later, then once every 3 years. • Dogs first vaccinated when older than 4 months are revaccinated 3 years later, then every 3 years. • In healthy dogs, the protective immune response induced by the MLV and rCDV vaccines is expected to last for at least 5 years. • The rCDV and MLV vaccines may be used interchangeably. • Vaccines must be used within one hour of reconstitution, as they lose virulence rapidly. Ideally, they are administered to the patient immediately after reconstitution.

Canine vaccination guideline for general veterinary practice

(modified from AAHA 2011 canine vaccination guideline)

Canine vaccine	Initial puppy vaccine (< 16 weeks)[1]	Initial vaccination for dogs older than 16 weeks	Revaccination
*Canine Adenovirus-2[3] (CAV2) MLV	Give one dose every 3-4 weeks between the ages of 6 and 16 weeks.	One dose is considered protective and acceptable.	• Puppies last vaccinated at 16 weeks of age should be revaccinated one year later, then once every 3 years. • Dogs first vaccinated when older than 4 months are revaccinated 3 years later, then every 3 years. • In healthy dogs, the protective immune response induced by the CAV2 vaccine is expected to last for at least 7 years. • Vaccine must be used within one hour of reconstitution, as it loses virulence rapidly. Ideally, it is administered to the patient immediately after reconstitution.

Canine vaccination guideline for general veterinary practice

(modified from AAHA 2011 canine vaccination guideline)

Canine vaccine	Initial puppy vaccine (< 16 weeks)[1]	Initial vaccination for dogs older than 16 weeks	Revaccination
*Rabies 1-year Killed	• Administer one dose as soon as possible after 12 weeks of age. • The dog is considered to have a protective titer 28 days after vaccination.	• One dose is considered protective and acceptable. • The dog is considered to have a protective titer 28 days after vaccination.	• Revaccinate one year after the first dose, then annually per law. • Booster vaccinations given on schedule to previously vaccinated dogs are considered to be immediately protective, so there is no break in protective antibody titer. • Read vaccine label to determine whether to administer the vaccine IM or SQ. • The use of single-dose vials is recommended to reduce the risk of contamination and to ensure proper mixing and dosage of antigen and adjuvant.

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Canine vaccination guideline for general veterinary practice

(modified from AAHA 2011 canine vaccination guideline)

Canine vaccine	Initial puppy vaccine (< 16 weeks)[1]	Initial vaccination for dogs older than 16 weeks	Revaccination
*Rabies 3-year Killed	• Administer one dose as soon as possible after 12 weeks of age. • The dog is considered to have a protective titer 28 days after vaccination.	• One dose is considered protective and acceptable. • The dog is considered to have a protective titer 28 days after vaccination.	• Revaccinate one year after the first dose, regardless of the age of the dog at the time of the first vaccination. Thereafter, administer one dose every 3 years (with the “3-year” vaccine), or every one year (with the “1-year” vaccine), or according to law. • Booster vaccinations given on schedule to previously vaccinated dogs are considered to be immediately protective, so there is no break in protective antibody titer. • Read vaccine label to determine whether to administer the vaccine IM or SQ. • The use of single-dose vials is recommended to reduce the risk of contamination and to ensure proper mixing and dosage of antigen and adjuvant.

Canine vaccination guideline for general veterinary practice

(modified from AAHA 2011 canine vaccination guideline)

Canine vaccine	Initial puppy vaccine (< 16 weeks)[1]	Initial vaccination for dogs older than 16 weeks	Revaccination
Parainfluenza Virus (CPiV) MLV	Give one dose every 3-4 weeks between the ages of 6 and 16 weeks.	One dose is considered protective and acceptable.	• CPiV vaccine is available only in combination with other core vaccines. Follow vaccination instructions as for CDV, CPV2 and CAV2. • Note that vaccination with CPiV prevents the dog from becoming ill, but does not prevent the dog from becoming infected with the virus and from shedding the virus.
Bordetella bronchiseptica inactivated cellular antigen extract, for parenteral (SQ) administration	Administer first dose at 8 weeks and the second dose at 12 weeks of age.	Two doses, 2-4 weeks apart. A single dose will not immunize a seronegative dog.	• Annually, or more often in very high-risk environments in which animals are not protected by annual booster. • This vaccine is protective 7-10 days after the second vaccination. The second dose of the initial series should be administered at least 1 week prior to anticipated exposure (e.g., vaccinate 1-2 weeks before exposing the dog to a kennel or other populations of dogs).

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Canine vaccination guideline for general veterinary practice

(modified from AAHA 2011 canine vaccination guideline)

Canine vaccine	Initial puppy vaccine (< 16 weeks)[1]	Initial vaccination for dogs older than 16 weeks	Revaccination
Bordetella bronchiseptica live avirulent bacteria, intranasal administration	• Administer one dose at the same time as the core vaccines. • The initial intranasal dose may be administered as early as 3-4 weeks of age when exposure risk is high.	One dose	• Annually, or more often in very high-risk environments in which surrounding animals are not protected by annual booster. • May see transient coughing, sneezing or nasal discharge. • Main advantage of intranasal over parenteral vaccine is the speed of onset of protective immunity within 72 hours. Advised if there is insufficient time for a parenteral vaccination to take effect, e.g., in a rescue / shelter situation (see next table). • Do not use the intranasal vaccine for parenteral injection. • No advantage to using the parenteral and intranasal vaccines together.

Canine vaccination guideline for general veterinary practice

(modified from AAHA 2011 canine vaccination guideline)

Canine vaccine	Initial puppy vaccine (< 16 weeks)[1]	Initial vaccination for dogs older than 16 weeks	Revaccination
Borrelia burgdorferi (Lyme disease) Killed or whole cell bacterin, or rLyme:rOspA	• Administer 1 dose at 12 weeks of age or later. Repeat 2-4 weeks later. • Do not administer to puppies younger than 12 weeks of age.	Two doses, 2-4 weeks apart. A single dose will not immunize a seronegative dog.	• Annually, or just before the regional tick season. • Note that tick control measures must be used to protect the dog in addition to vaccination. • Recommended to be used only for animals with known risk of exposure or in areas where Lyme's disease is endemic.
Leptospira interrogans killed whole cell or subunit bacterin. Contains 4 serovars: [4] canicola, icterohemorrhagiae, grippotyphosa, pomona.	• Administer 1 dose at 12 weeks of age or later. Repeat 2-4 weeks later. • Do not administer to puppies younger than 12 weeks of age.	Two doses, 2-4 weeks apart. A single dose will not immunize a seronegative dog.	• Annual booster. • Dogs should be vaccinated only if there is a reasonable risk of exposure. • The previous 2-way bacterin is no longer recommended.

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Canine vaccination guideline for general veterinary practice

(modified from AAHA 2011 canine vaccination guideline)

Canine vaccine	Initial puppy vaccine (< 16 weeks)[1]	Initial vaccination for dogs older than 16 weeks	Revaccination
Canine influenza vaccine Killed	Administer one dose after 6 weeks of age, and a second dose 2-4 weeks later.	Two doses, 2-4 weeks apart. A single dose will not immunize a seronegative dog.	• Do not vaccinate unless both doses can be given. • Annual revaccination • Use this vaccine in areas in which CIV is endemic, or if animals are transported to or from endemic areas. In most cases, it is not considered a core vaccine.
Canine Coronavirus (CCoV)	Not recommended, as vaccination does not reduce disease caused by CCoV or CPV-2.		

Canine vaccination guideline for shelter-housed dogs

(modified from AAHA 2011 canine vaccination guideline)

Canine vaccine	Initial vaccination[1]	Revaccination & Comments
*CDV + CAV2 + CPV2 + CPiV Modified-live vaccines (Do not use killed/ inactivated vaccines.)[2, 3]	• For all dogs older than 4 weeks of age, administer a single dose immediately before or at the time of admission. • Exceptions to this are if 1) there is written evidence that the dog has been appropriately and currently vaccinated, or 2) the dog is demonstrated to have protective levels of circulating antibody against CDV and CPV2.	• Puppies younger than 18 weeks: vaccinate every 2 weeks until 18-20 weeks of age. • Dogs older than 5 months of age: revaccinate at one year of age. • Thereafter, revaccinate all dogs every 3 years, per standard recommendation.
Measles vaccine (available in a 4-way combination with CDV+CAV2+CPiV or in a 2-way combination with CDV.) Administered IM only.	• Administer one dose between 6-12 weeks of age. • Follow every 2-4 weeks with CDV vaccine.	• MV provides temporary immunization to puppies against CDV in the presence of maternal antibodies, and has been shown to be effective 2 weeks earlier than the modified-live CDV vaccine. It may be used in environments of high risk for CDV infection for puppies who may have circulating maternal antibodies (younger than 4 months old). • This vaccine must be administered IM, and within 1 hour of reconstitution.

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Canine vaccination guideline for shelter-housed dogs

(modified from AAHA 2011 canine vaccination guideline)

Canine vaccine	Initial vaccination[1]	Revaccination & Comments
* *Bordetella bronchiseptica* Killed + CPiV-MLV Intranasal	Administer one dose immediately before or at the time of admission, as early as 3-4 weeks of age.	• Dogs younger than 6 weeks: revaccinate after 6 weeks of age, at least 2 weeks after the first dose. Dogs older than 6 weeks: revaccinate every 6-12 months. • Onset of protective immunity with the intranasal vaccine is within 72 hours. • Vaccination will reduce the severity of disease, but will not protect against infection or shedding of the respiratory disease complex. • This intranasal vaccine is recommended over the parenteral vaccine due to the more rapid onset of protective immunity. • The intranasal vaccine must be administered only intranasally. Do not inject IM or SQ.
Bordetella bronchiseptica parenteral (SQ) vaccine	Administer one dose at the time of admission. Repeat 2 weeks later. A single dose will not immunize a seronegative dog.	• Regardless of the dog's age, at least 2 vaccinations must be administered, 2 weeks apart, to induce immunity. A single dose will not immunize a seronegative dog. • Dogs who have received two vaccinations within the past 12 months require only one vaccination. • Parenteral B. *brochiseptica* vaccine is used only if the intranasal vaccine is not feasible. At least two doses, 2 weeks apart, are necessary to induce immunity; immunity is expected 7-10 days after the second dose. • Do not administer this product intranasally.

Canine vaccination guideline for shelter-housed dogs

(modified from AAHA 2011 canine vaccination guideline)

Canine vaccine	Initial vaccination[1]	Revaccination & Comments
*Rabies 1-year killed	• Administer one dose at the time of discharge from the shelter for all dogs 12 weeks of age or older. • If a longer stay at the shelter is anticipated, vaccinate on arrival at the shelter.	• Revaccinate one year after the initial vaccination, then every 1 or 3 years, per State, provincial, and/or local law. • The use of single-dose vials is recommended to reduce the risk of contamination and to ensure proper mixing and dosage of antigen and adjuvant.
Canine influenza vaccine (CIV) killed	Administer one dose after 6 weeks of age, and a second dose 2-4 weeks later.	• Seronegative dogs must receive two doses, 2-4 weeks apart. A single dose will not immunize a seronegative dog. • Do not vaccinate unless both doses can be given. • Use this vaccine in areas in which CIV is endemic, or if animals are transported to or from endemic areas. In most cases, it is not considered a core vaccine.

1: A dose of vaccine consists of the same volume for a puppy as for an adult dog. Vaccine doses should not be halved or otherwise reduced for young animals or small individuals.

2: rCDV (recombinant CDV vaccines) may be used interchangeably with MLV-CDV vaccine. Recent studies have shown that rCDV vaccines are more likely to mount an immune response in puppies who have passively-acquired maternal antibody. Recombinant vaccines also appear to generate a cellular immune response that is not seen with MLV or killed vaccine, and that is not reflected in measurement of a standard antibody titer. The drawback of recombinant vaccines for use in field situations is that they may be more expensive than MLV vaccines. Moreover, most CDV vaccines come in combination with the other core vaccines, and vaccinating for each individually increases the expense even further.

3: Vaccination with CAV-1 is not recommended due to significant risk of "hepatitis blue-eye" reactions. CAV-2 vaccines effectively cross-protect against CAV-1 and are much safer.

4: Note that dominant wild-type *Leptospira serovars* to which animals are exposed may vary regionally. The four that are listed here may not be those most relevant in some areas.

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Feline vaccination guideline for general veterinary practice

(modified from AAFP 2013 vaccination guideline)

Feline vaccine	Initial kitten vaccine (< 16 weeks)[5]	Initial vaccination for cats older than 16 weeks	Revaccination & Comments
* Feline Panleukopenia + Herpesvirus-1 + Calicivirus (FPV, FHV-1, FCV) MLV, SQ injection	Administer first dose as early as 6 weeks of age. Repeat every 3-4 weeks until 16-20 weeks of age.	Administer 2 doses, 3-4 weeks apart.	• Revaccinate 1 year after initial series, thereafter every 3 years. • Use parenteral vaccine rather than intranasal, particularly in high-risk environments. Do not use parenteral vaccines intranasally or vice versa. • Vaccination against FPV should protect cats very well. Cats vaccinated against respiratory infections may still become ill, but the infection will be relatively mild and shedding of virus reduced.
* Rabies killed or recombinant SQ or IM, depending on law	• Administer 1 dose as early as 12 weeks of age. • The kitten is considered to have a protective titer 28 days after vaccination.	• Administer 1 dose. • The cat is considered to have a protective titer 28 days after vaccination.	• Revaccinate 1 year later. Subsequently, revaccinate every 1-3 years as required by law • The AAFP does not consider rabies a core vaccine for cats. However, rabies vaccination protocols for cats must be followed according to local law. In areas where rabies is endemic, particularly in dogs, it is advisable to vaccinate cats with rabies as a core vaccine.

Feline vaccination guideline for general veterinary practice

(modified from AAFP 2013 vaccination guideline)

Feline vaccine	Initial kitten vaccine (< 16 weeks)[5]	Initial vaccination for cats older than 16 weeks	Revaccination & Comments
Feline Leukemia virus (FeLV)[6] killed or recombinant SQ injection	• Administer first dose as early as 8 weeks of age. • Repeat 3-4 weeks later.	Administer 2 doses, 3-4 weeks apart.	• Cats should be tested and confirmed FeLV-negative before vaccination. • Revaccinate 1 year after the initial series. Thereafter, revaccinate cats at low risk of infection every two to three years; cats at high risk of infection annually. • Kittens younger than 1 year of age should be vaccinated as a matter of routine, provided that they test FeLV-negative.
Feline Immunodeficiency Virus (FIV)[7] killed, SQ injection	Three doses are required: initial dose as early as 8 weeks of age; two subsequent doses in 2-3 week intervals.	Three doses are required, each 2-3 weeks apart.	• May be recommended for outdoor cats or for cats living with FIV-positive individuals. • Vaccination is controversial. See Footnote 7.
Clamydophila felis[8] avirulent live or killed SQ injection	• Administer at the time of admission, as early as 9 weeks of age. • Administer the second dose 3-4 weeks later if the kitten is still in the shelter.	• Administer at the time of admission. Repeat 3-4 weeks later if the cat is still in the shelter.	• Neither vaccination or natural infection induce a sterile immunity. Cats may continue to shed *C. felis* for months. • Ocular inoculation with vaccine will result in typical clinical disease.

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Feline vaccination guideline for general veterinary practice

(modified from AAFP 2013 vaccination guideline)

Feline vaccine	Initial kitten vaccine (< 16 weeks)[5]	Initial vaccination for cats older than 16 weeks	Revaccination & Comments
Bordetella bronchiseptica[8] avirulent live organism intranasal	• Administer a single dose as young as 4 weeks of age, at the time of admission to the shelter.	• Administer a single dose at the time of admission to the shelter.	• Upper respiratory disease following intranasal vaccination may be impossible to distinguish from active infection. • Onset of immunity may begin as early as 3 days after inoculation. • Do not use the canine *Bordetella* vaccines in cats.
Feline Corona Virus (Feline Infectious Peritonitis)	Vaccination against feline coronavirus is generally not recommended.		

Feline vaccination guideline for shelter-housed cats

(modified from AAFP 2013 vaccination guideline)

Feline vaccine	Initial kitten vaccine (< 16 weeks)[5]	Initial vaccination for cats older than 16 weeks	Revaccination & Comments
*Panleukopenia + Herpesvirus-1 + Calicivirus (FPV, FHV-1, FCV) MLV, SQ injection	• Administer first dose at time of admission, as early as 4-6 weeks of age. • Repeat every 2-3 weeks until 16-20 weeks of age if still in the shelter. • In high-risk environments, kittens should be vaccinated at 4 weeks, and every 2 weeks thereafter until 16-20 weeks of age or rehomed.	Administer 1 dose at time of admission. Repeat in 2-3 weeks.	• Do not administer to kittens younger than 4 weeks old due to risk of cerebellar hypoplasia or clinical panleukopenia. • Use MLV vaccine in healthy, non-pregnant kittens & cats. For pregnant queens, risk of exposure and protection of the shelter environment must be weighed against the risk to the fetuses. • In group-housed, long-term shelters in which vaccinated cats become ill with calicivirus, a multivalent or different strain of calicivirus vaccine may be beneficial.
Herpesvirus-1 + Calicivirus (FHV-1, FCV) MLV, intranasal	• Administer first dose at time of admission, as early as 4-6 weeks of age. • Repeat every 2-3 weeks until 16-20 weeks of age if still in the shelter.	Administer 1 dose at time of admission. Repeat in 2-3 weeks.	• Onset of protection following intranasal vaccination may begin within 4-6 days, and may therefore provide more rapid protection in a high-risk situation. • Transient, mild signs of upper respiratory disease may develop following intranasal vaccination. • If intranasal vaccine is used to control upper respiratory infection, all cats must receive parenteral FPV vaccine at the same time.

Feline vaccination guideline for shelter-housed cats

(modified from AAFP 2013 vaccination guideline)

Feline vaccine	Initial kitten vaccine (< 16 weeks)[5]	Initial vaccination for cats older than 16 weeks	Revaccination & Comments
*Rabies killed or recombinant SQ or IM, depending on law	• Administer one dose to cats 3 months of age or older at the time of discharge from the shelter. • Cats who will remain in the shelter long-term may be vaccinated at time of admission if rabies exposure is considered a potential risk in the shelter environment.	• Administer one dose at the time of discharge from the shelter. • Cats who will remain in the shelter long-term may be vaccinated at time of admission if rabies exposure is considered a potential risk in the shelter environment.	• Revaccinate one year after the initial vaccination. • Subsequent revaccination schedule depends on the risk exposure and local laws. Generally, this is every 1-3 years.
FeLV[6] killed or recombinant SQ injection	• Vaccinate as early as 8 weeks of age for group-housed kittens. • Repeat vaccination 2-3 weeks later.	• Vaccinate on arrival all group-housed cats. • Revaccinate 3-4 weeks after the first vaccine.	• FeLV vaccine is recommended for cats in long-term shelters or in shelters that house unrelated cats in groups. • Individually housed shelter cats may not require vaccination, provided that there are good biosecurity protocols. • Vaccination should be done in conjunction with a program of testing and segregating infected cats (see Footnote 6).

Feline vaccination guideline for shelter-housed cats

(modified from AAFP 2013 vaccination guideline)

Feline vaccine	Initial kitten vaccine (< 16 weeks)[5]	Initial vaccination for cats older than 16 weeks	Revaccination & Comments
Clamydophila felis[8] avirulent live or killed SQ injection	• Administer at the time of admission, as early as 9 weeks of age. • Administer the second dose 3-4 weeks later if the kitten is still in the shelter.	• Administer at the time of admission. Repeat 3-4 weeks later if the cat is still in the shelter.	• Neither vaccination or natural infection induce a sterile immunity. Cats may continue to shed *C. felis* for months. • Ocular inoculation with vaccine will result in typical clinical disease.
Bordetella bronchiseptica[8] avirulent live organism intranasal	• Administer a single dose as young as 4 weeks of age, at the time of admission to the shelter.	• Administer a single dose at the time of admission to the shelter.	• Sneezing or coughing after intranasal vaccination may be impossible to distinguish from active infection. • Onset of immunity may begin as early as 3 days after inoculation. • Do not use the canine *Bordetella* vaccines in cats.
Feline immunodeficiency virus (FIV) and feline coronavirus (feline peritonitis) (FCoV, FIP)	Vaccination not recommended in shelters. Follow isolation and management procedures to protect uninfected cats.		

5: A dose of vaccine consists of the same volume for a kitten as for an adult cat. Vaccine doses should not be halved or otherwise reduced for young animals or small individuals.

6: Cats should be tested and confirmed FeLV negative before vaccination. FeLV vaccine is recommended only for FeLV-negative cats who go outdoors, who come into contact with FeLV infected cats, or who live in an environment in which cats frequently are brought into contact with new cats of unknown FeLV status. FeLV vaccination is highly recommended for all kittens, but boosters are not recommended for cats who are strictly confined indoors and who do not come into contact with new or potentially FeLV-positive cats. FeLV-infected cats should be kept indoors and isolated from susceptible cats, and from cats who may carry other infectious pathogens. FeLV-infected cats should be vaccinated with core vaccines, but these cats may not mount an adequate immune response to protect them from challenge.

7: FIV vaccine should be used only for cats who are at high risk of infection. Cats at high risk include outdoor cats who fight with other cats and cats who live with FIV-infected cats. Vaccination with FIV induces production of antibodies that are indistinguishable from antibodies produced against naturally-acquired infection. Therefore, vaccination will interfere with all antibody-based diagnostic tests for at least 1 year following vaccination. Cats should be tested and confirmed FIV negative before vaccination. Note that kittens may carry maternally-derived antibodies until ca. 12 weeks of age. FIV-infected cats should be kept indoors and isolated from susceptible cats, and from cats who may carry other infectious pathogens. FIV-infected cats are able to mount an immune response to vaccination with core vaccines until the terminal stages of FIV-associated disease, but the immune response may be delayed or diminished compared with FIV-negative cats.

8: Routine vaccination against *Chlamydophila felis* and *Bordetella bronchiseptica* is not recommended, even for shelter cats. The association between either of these infections and respiratory disease is inconsistent. Vaccination against these pathogens is reserved for cases where it is supported by laboratory diagnostics. *B. bronchiseptica* vaccination may also be considered where there is potential exposure of cats to dogs in shelters with a "kennel cough" problem. Note that even cats vaccinated with these organisms may shed the pathogen for weeks or months, and may pose a risk to other susceptible animals. The objective of vaccination is not to prevent disease, but to lessen the clinical impact of infection.

Appendix 4: Anti-parasitic drugs

A faecal parasite examination should be done every 6-12 weeks (depending on climate and living conditions), and the animal treated as necessary. In shelters, all animals should be treated for internal and external parasites on arrival. A regular dosing schedule with rotating use of different anthelminitics should be part of the standard shelter operating protocol, with periodic faecal checks on a random selection of animals representing different shared enclosures.

Compliance by animal guardians with preventive veterinary protocols like parasite control may be challenging, particularly in areas in which people have little access to veterinary care. In these areas, the project should include strategies for improving access to primary veterinary care, and for educating animal guardians in the importance of preventive health care for their animals. Zoonotic risk management of parasites is also a consideration, particularly for the protection of children. Preventive veterinary health care is essential for the health and welfare of the individual animal, for other animals in the community, and for the safety and welfare of people in the community.

The following is a summary of common drugs available for treatment of endoparasites and ectoparasites. The list is comprehensive, but not exhaustive. Trade names of drugs will differ among countries, but trade names of some of the common internationally-distributed products (often combinations of drugs) have been listed here.

PO = per os (oral) **SQ** = subcutaneous
IM = intramuscular **SID** = once per day (once every 24 hours) **BID** = twice per day (every 12 hours)
q = every (e.g., q8 hrs = every 8 hours)
Monthly = once per month

Topical or **Topical spot-on** = Solution that is to be applied to the skin of the dog or cat, usually on the dorsal neck between the shoulder blades where the animal cannot reach it with the tongue. Care should be taken to prevent the compound from getting on the skin of the person applying the medication.

Roundworms refers to nematodes, including *Toxocara, Ascaris*, and lungworms *(Aelurostrongylus, Capillaria, Filaroides)*. Does not necessarily include canine whipworms *(Trichuris vulpis)*. **Hookworms**: *Ancylostoma* and *Uncinaria spp*. **Tapeworms/cestodes**: *Dipylidium, Taenia, Echinococcus, Diphilobothrium, Spirometra*.

Drug or Product	Application	Dose & comments
Adam's flea products (Farnam pet products) Pyrethrins, piperonyl butoxide, (S)-methoprene, N-octyl bicycloheptene dicarboximide	Topical spray, kills & repels: • fleas: adults, eggs & larvae • ticks • lice • mosquitoes, gnats, flies	• Dogs & cats > 3 months old. • Avoid use on cats (pyrethrins) • Bedding • Washes off in rain and water.
Advantage® (Bayer) Imidacloprid	• Monthly topical spot-on, flea prevention treatment. • Fleas: adults & larvae	• Dogs & cats > 7 months old • Prevents fleas from biting, and kills those that do bite. The former property makes it useful in management of flea allergies. • Not effective against ticks
Advantage-Multi® / Advocate® (Bayer) Imidacloprid + moxidectin	Monthly topical spot-on: • fleas: adult & larvae • heartworm prevention • hookworm • roundworm • whipworm (dogs) • ear mites (cats)	• For dogs and cats > 1.5 kg and > 9 weeks old • Different formulations for dogs and cats, use per label instructions. • Not effective against demodectic mange, although package suggests use for this condition. • Moderately effective against sarcoptic mange in dogs, but use only if ivermectin or selamectin are not options. • Do not bathe after application.
Advantix K9 (Bayer) Imidacloprid + permethrin	• Monthly topical spot-on for dogs only • Prevention of fleas (adult & larvae), ticks, mosquitoes	• Do not use in cats (permethrins) • Topical spot-on, applied once per month, dose per label instructions.

Drug or Product	Application	Dose & comments
Albendazole	• Roundworms (incl. lungworms) • Paragonimus (lung flukes) • *Capillaria* (dogs) • *Giardia*	• Dogs: *Filaroides* (lungworms): 50 mg/kg BID, PO x 5 days, repeat in 3 weeks. • Dogs: *Capillaria*: 50 mg/kg BID, PO x 14 days • *Paragonimus*: 25-50 mg/kg BID, PO for 14-21 days. • *Giardia*: 25-50 mg/kg BID x 5 days • Liver flukes in cats: 50 mg/kg SID, PO until ova are gone. • May cause bone marrow suppression in dogs & cats.
Atovaquone + azithromycin	• Antiprotozoal for treatment of babesiosis in dogs	• Atovaquone 13.3 mg/kg PO q8 hr + azithromycin 10 mg/kg PO SID for 10 days • See also imidocarb dipropinate and diminazene aceturate
Capstar® (Novartis) Nitenpyram	• Kills adult fleas for up to 24 hours. • For treatment of dogs and cats who are infested with adult fleas, as a first step in flea control	• Oral tablet, dose per package instructions • For dogs and cats > 4 weeks and > 1kg. • Works within 30 minutes. Within 4 hours, kills all the fleas on the animal. Lasts for 24 hours in killing new fleas.
Certifect® (Merial) Fipronil, S-methoprene, amitraz	• Monthly topical spot-on • Fleas: adults & larvae. • Ticks (stronger than Frontline)	• Dogs only • Waterproof formulation • Topical spot-on, applied once per month, dose per label instructions.

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Drug or Product	Application	Dose & comments
Cestex® (Pfizer)	Dog and cat tapeworms: *Dipylidium and Taenia*	See Epsiprantel
Comfortis® (Eli Lilly) Spinosad	• Monthly flea prevention • Kills adult fleas only.	• Dogs and cats > 14 weeks. Cats > 1kg, dogs > 1.5 kg. • Oral: dose per package instructions (23-45 mg/kg). Separate formulations for dogs and cats.
Dichlorvos	Roundworms	• Dog & cat: 11 mg/kg PO • Organophosphate drug; dose carefully. Preferable to use a newer, safer compound.
Diethylcarbamazine	• Daily oral heartworm preventative • Ascarids	• Dog 6.6 mg/kg PO once per day for heartworm prevention. Now rarely used; replaced with monthly heartworm preventation options. • Ascarids, dogs & cats: 55-110 mg/kg PO, once. Repeat in 21 days. • Note that there are newer, more broad-spectrum drugs now available for treatment of intestinal parasites.
Diminazene aceturate	• Antipropozoal • *Babesia* • *Cytauxzoon* • *Trypanosoma*	• *Babesia & Trypanosoma* (dogs): 3.5-5 mg/kg IM. Repeat in 24 hrs. Risk of neurotoxicity higher if total dose is > 7 mg/kg • *Cytoxzoon* (cats): 3-5 mg/kg IM, once, or 2 mg/kg IM and repeat in 1 week. • May not clear infections completely; consider imidocarb dipropionate.
Doramectin	Use and dose same as ivermectin	See ivermectin

Drug or Product	Application	Dose & comments
Drontal™ 20 mg Praziquantel + 230 mg Pyrantel embonate per tablet	• Roundworms • Hookworms • Tapeworms	• Cats > 4 weeks, > 700g: dose per label instructions. One tablet per 4 kg: 20 mg praziquantel + 230 mg pyrantel embonate. • Dogs: use Drontal-Plus / Drontal Allwormer.
Drontal-Plus™ or Drontal Allwormer febantel + pyrantel pamoate + praziquantel	• Roundworms • Hookworms • Whipworms • Tapeworms	• Dogs > 3 weeks, > 1kg: dose per label instructions. One tablet per 10 kg: 50 mg praziquantel + 49.8 mg pyrantel embonate + 250 mg febantel. • Use of Drontal-Plus is not recommended in cats, as febantel is not well tolerated and often causes vomiting in cats.
Effipro® (Virbac) Fipronil	• Monthly spot-on flea & tick control • Kills adult fleas & ticks (does not kill eggs & larvae)	• For cats > 8 weeks & 1 kg. • Dogs > 8 weeks & 2 kg • Kills adult fleas within 24 hrs and ticks within 48 hours after they bite. • Does not prevent fleas & ticks from attaching or biting the animal, hence not effective for flea allergy management.
Epsiprantel Cestex® (Pfizer)	Dog and cat tapeworms: *Dipylidium* and *Taenia*	• Dogs and cats > 7 weeks old • Dog: 5.5 mg/kg PO • Cat: 2.75 mg/kg PO

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Drug or Product	Application	Dose & comments
Fenbendazole (Panacur®)	• Ascarids • Hookworms • Whipworms • *Taenia* spp. • Tapeworms • *Giardia* • Drug of choice for giardiasis.	• Requires dosing for 3 consecutive days, 50 mg/kg SID. Repeat in 3 wks. • UK allows 100 mg/kg, once. Repeat in 3 wks. • Treatment of *Capillaria* may need to be extended to 10 days. • For pregnant bitches: 25 mg/kg SID from day 40 gestation until 2 days post-whelping (approximately 25 days). • Lungworms: treat cats 5 consecutive days, dogs 7 days • *Giardia*: 50 mg/kg PO once daily for 5 days.
Fipronil	Topical spot-on Kills adult fleas and ticks	• For cats > 8 weeks & 1 kg. • Dogs > 8 weeks & 2 kg • Kills adult fleas within 24 hrs and ticks within 48 hours after they bite. • Does not prevent fleas and ticks from attaching or biting the animal, hence not effective for flea allergy management. • See Effipro, Frontline, Frontline-Plus, Certifect.
Flubendazole	• Roundworms • Hookworms • Whipworms (*Trichuris vulpis*; dogs) • Tapeworm (*Taenia pisiformis*; dog & cat)	• Roundworms & hookworms: 22 mg/kg PO SID x 2 days. Repeat in 3 weeks. • *Trichuris vulpis* (dogs) & *Taenia pisiformis* (dogs & cats): 22 mg/kg SID x 3 days. Repeat in 3 weeks.

Drug or Product	Application	Dose & comments
Frontline® (Merial) Fipronil	• Monthly flea & tick control.	• See Fipronil • Available as spray or spot-on • Washes off with water. Do not bathe for 3 days after application.
Frontline-Plus® (Merial) Fipronil + S-methoprene	• Monthly flea & tick control • Fleas: adults & larvae & eggs	• Dogs & cats: use per label instructions. • Addition of S-methoprene to fipronil makes this product effective against immature stages of fleas, hence more effective in controlling flea populations in the environment. • Available as spray or spot-on • Do not bathe for 3 days after application.
Furazolidone	• Antiprotozoal and antibacterial • Second-choice drug against protozoa. • *Giardia*, *Trichomonas*, coccidia • Some strains of *E coli*, *Enterobacter*, *Campylobacter*, *Salmonella*, *Shigella*, *Vibrio cholerae*	• *Giardia*, dogs & cats: 4 mg/kg PO BID x 7-10 days • Coccidiosis, dogs & cats: 8-20 mg/kg SID PO x 7 days • Amoebic colitis, dogs & cats: 2.2 mg/kg PO q8 hrs x 7 days
Heartgard® (Merial) Ivermectin, low-dose	• Monthly prevention of heartworms • Note: not effective against intestinal parasites, sarcoptic mange or demodecosis.	• Dogs and cats: oral, dose per package instructions. • Ivermectin dose in Heartgard and Heartgard-Plus is 0.006-0.012 mg/kg. Note that this is a heartworm prevention dose only. At this dose, it is not effective against intestinal parasites, *Sarcoptes*, *Demodex* or ear mites. See Ivermectin for doses appropriate against these pathogens.

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Drug or Product	Application	Dose & comments
Heartgard-Plus® (Merial)	• Monthly prevention of: • Heartworm • Hookworms • Roundworms • The low dose of ivermectin in this product makes it ineffective against sarcoptic mange or *Demodex*.	• Dogs and cats: oral, dose per package instructions. • Ivermectin dose in Heartgard and Heartgard-Plus is 0.006-0.012 mg/kg. Note that this is a heartworm prevention dose only. At this dose, it is not effective against intestinal parasites, Sarcoptes, Demodex or ear mites. See Ivermectin for doses appropriate against these pathogens. • The addition of pyrantel in this product makes Heartgard-Plus® effective against hookworms and roundworms.
Imidacloprid	• Monthly topical spot-on, flea prevention treatment. • Fleas: adults & larvae	• See Advantage, Advantage-Multi, Advantix K9
Imidocarb dipropinate Imizol® (Schering-Plough)	• Antiprotozoal for treatment of babesiosis in dogs and *Cytauxzoon felis* in cats	• Dogs: 6.6 mg/kg IM or SQ, one dose. Repeat in 14 days. • Cats: 5 mg/kg IM, one dose. Repeat in 14 days. • Supportive care during treatment is essential.
Interceptor® (Novartis) milbemycin oxime	• Monthly heartworm prevention	• see milbemycin oxime

Drug or Product	Application	Dose & comments

Ivermectin Heartgard®, Heartgard-Plus®, Ivomec® (Merial)	• Heartgard and Heartgard-Plus are used for the prevention of heartworm in heartworm-negative dogs.
	• For ivermectin-sensitive dogs, use another options for heartworm prevention, such as milbemycin.
	• Ivermectin (1% solution): roundworms, lungworms, *Demodex* (need high dose), sarcoptic and otodectic mange
		• See Heartgard® and Heartgard-Plus®
		• Do not give ivermectin or doramectin to collies, sheepdogs, other herding breeds or mixed-breed dogs of these breeds, or to other dogs with known ivermectin sensitivity (MDR1 gene defect). If in doubt, use a different drug.
		• Dogs must be checked and ascertained to be free of adult heartworms prior to giving avermectins. Adult heartworms produce large numbers of microfilaria. Treatment of heartworm-positive dogs with ivermectin may cause mass death of microfilariae in the bloodstream, which in turn may precipitate a shock-like reaction in the dog.
		• Avermectins do not kill adult heartworms. Dogs with adult heartworms must be treated with melarsomine under controlled conditions and appropriate pre- and post-treatment adjunct therapy.
		• Dog doses
		  • Heartworm prevention: 0.006-0.012 mg/kg PO once per month (e.g. Heartgard)
		  • Ectoparasiticide (*Sarcopes, Otodectes*): 0.3 mg/kg SQ or PO once. Repeat once weekly for at least 3-4 weeks until 3 consecutive skin scrapings, 1 week apart, are found negative. See mange treatment protocols.
		  • *Demodex*: 0.3-0.6 mg/kg SID PO or SQ until 3 consecutive skin scrapings, 3 weeks apart, are found negative. Ramp up to full dosage gradually at the beginning of treatment, and taper off dosage gradually at the end of treatment: do not start and end abruptly. At high doses, monitor closely for signs of toxicity. See demodecosis treatment protocol.
		• Cats: Heartworm prevention: 0.024 mg/kg PO once per month. Otodectes as for dogs.
		• Roundworms & hookworms, dogs & cats: 0.2-0.4 mg/kg PO or SQ once. Repeat in 3 wks.
		• Lungworm: 0.4 mg/kg SQ, once

Drug or Product	Application	Dose & comments
Ivermectin Heartgard®, Heartgard-Plus®, Ivomec® (Merial) *(Continued from page 69)*	• Heartgard and Heartgard-Plus are used for the prevention of heartworm in heartworm-negative dogs. • For ivermectin-sensitive dogs, use another options for heartworm prevention, such as milbemycin. • Ivermectin (1% solution): roundworms, lungworms, *Demodex* (need high dose), sarcoptic and otodectic mange	end of treatment: do not start and end abruptly. At high doses, monitor closely for signs of toxicity. See demodecosis treatment protocol. • Cats: Heartworm prevention: 0.024 mg/kg PO once per month. *Otodectes* as for dogs. • Roundworms & hookworms, dogs & cats: 0.2-0.4 mg/kg PO or SQ once. Repeat in 3 wks. • Lungworm: 0.4 mg/kg SQ, once
Levamisole	• Roundworms • Lungworms	• Used primarily in livestock, rarely in dogs & cats now. • Dogs, lungworms: 7-10 mg/kg SID, PO x 7 days (*Capillaria*) or 45 days (*Filaroides*). • Cats, lungworms: various dosing regimes, highly variable. Refer to Plumb's. • Has immune stimulant properties • Supportive care during treatment is essential.
Lufenuron	• Monthly control of flea eggs and larval development	• see Program, Sentinel.
Meglumine antimoniate	• Leishmaniasis in dogs	• May combine with allopurinol. • Minimum dose: 100 mg/kg SQ SID 3-4 weeks; better if 4-6 wks • Adding allopurinol at 20-40 mg/kg PO SID for several months may reduce relapse rate.

Drug or Product	Application	Dose & comments
Melarsomine	• Treatment of adult heartworms in dogs	• Careful pre-treatment clinical evaluation, pre-treatment medications and strict exercise restriction are necessary • Refer to full preparation and monitoring protocols. Three different dosing options are recommended. Most commonly used protocol: 2.5 mg/kg IM on Day 1. Wait 1-3 months, then deliver 2 more doses, 24 hours apart.
Metronidazole	• Antiprotozoal: *Giardia, Trichomonas, Entamoeba, Balantidium.* • Anaerobic bacteria	• *Giardia*, dogs & cats: 15-30 mg/kg PO once daily x 5-7 days • *Entamoeba*, dogs & cats: 25 mg/kg BID PO x 8 days
Lufenuron	• Monthly control of flea eggs and larval development	• see Program, Sentinel.
Milbemax® (Novartis) Milbemycin oxime + praziquantel	• Monthly prevention against heartworm, intestinal nematodes & tapeworms for dogs and cats	• For dogs more than 2 weeks old and more than 0.5 kg. Dosed at 0.5 – 2.5 mg/kg milbemycin + 5-25 mg/kg praziquantel. • For cats and kittens: 2-4 mg/kg milbemycin + 5-10 mg/kg praziquantel.

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Drug or Product	Application	Dose & comments
Milbemycin oxime	• Monthly heartworm prevention • Treatment of sarcoptic or demodectic mange or cheyletiellosis in ivermectin-sensitive breeds of dogs	• Dogs, heartworm prevention: 0.5-2.5 mg/kg PO once per month • Cats, heartworm prevention: 2-4 mg/kg PO once per month • May be used in ivermectin-sensitive dogs, but dose must be followed precisely. Smaller margin of safety in these dogs than in individuals who are not hypersensitive to ivermectin. • May be moderately effective against sarcoptic mange or cheyletiellosis if ivermectin or selamectin are not options. 2 mg/kg PO once every 7 days for 3 doses. • May be effective against demodectic mange in ivermectin-sensitive breeds. 1-2 mg/kg PO SID 30-60 days. • See Interceptor (milbemycin oxime) • See Sentinel (Milbemycin oxime + lufenuron) • See Trifexis (milbemycin oxime + spinosad) • See Milbemax (milbemycin oxime + praziquantel)
Moxidectin	• Heartworm prevention • Hookworms • Roundworms	• See Proheart and Advantage-Multi / Advocate • May be used in ivermectin-sensitive dogs, but dose must be followed precisely. Smaller margin of safety in these dogs than in individuals who are not hypersensitive to ivermectin.

Drug or Product	Application	Dose & comments
Nitenpyram	• Adult fleas	• See Capstar®
Oxfendazole	• Lungworm in dogs (*Filaroides*)	• 10 mg/kg SID PO x 28 days
Permethrin	• Ectoparasite control in dogs	• See also Advantix K9 • Do not use in cats.
Piperazine	• Roundworms & hookworms	• Roundworms, dogs & cats: 55-100 mg/kg PO, repeat 10 days later. • Hookworms, dogs & cats: 120-240 mg/kg
Profender™ (Bayer) Emodepside + praziquantel	• Topical spot-on for cats, monthly • Hookworms • Roundworms • Tapeworms	• Cats > 8 weeks old, > 1 kg. • Minimum dose: 3 mg/kg emodepside + 12 mg/kg praziquantel, topically
Program® (Novartis) lufenuron	• Monthly control of flea egg & larval development	• Dogs & cats > 6 weeks old • Dogs & cats: oral tablets, per package instructions, once per month. • Cats: oral suspension, per package instructions, once per month • Cats: 6-month injectable (do not use in dogs). • Insect growth regulator, stops flea eggs & larvae from developing. Works by transferring drug to the adult flea when it takes a blood meal from the dog or cat. Eggs produced subsequently by the flea are not viable. Does not prevent the flea from biting the dog or cat, hence not preventative against flea allergy dermatitis.

Drug or Product	Application	Dose & comments
ProHeart®6, ProHeart®12 (Pfizer) Guardian®SR, Moxidec®SR (Fort Dodge) Moxidectin	• Injectable, six- or twelve-month heartworm preventative for dogs • Hookworms	• Sustained-release moxidecin, administered once every 6 months (ProHeart-6) or 12 months (ProHeart-12) by SQ injection at 0.17 mg/kg. • Effective against heartworm microfilaria (heartworm preventative) and hookworms. • For dogs more than 6 months old.
Pyrantel pamoate	• Roundworms • Hookworms • Stomach worm (*Physaloptera*)	• Dogs and cats > 3 weeks old, 5-10 mg/kg PO, repeat in 3 weeks • See Drontal® (pyrantel + praziquantel) • See Drontal-Plus® (febantel + pyrantel pamoate + praziquantel) • See Heartgard-Plus® (ivermectin + pyrantel)
Pyrimethamine	• Antiprotozoal: *Toxoplasma, Hepatozoon, Neospora*	• Combined with sulfadiazine serves as an alternative to trimethoprim / sulfadiazine • Caution in cats: bone marrow suppression. • Dosing varies with pathogen & protocol
Quinacrine	• Second-choice drug for *Giardia, Leishmania*, coccidia	• Dogs: 6.6 mg/kg PO BID x 5 days • Cats: 9 -10 mg/kg PO SID x 5 days; for *Giardia* up to 10 days.
Revolution™ (Pfizer)	• Monthly topical spot-on for dogs and cats against ectoparasites & endoparasites and heartworm prevention.	• See selamectin

Drug or Product	Application	Dose & comments
Ronidazole	• Antiprotozoal for *Trichomonas foetus* infection in cats	• Cats: 30 mg/kg BID PO x 14 days
Selamectin Revolution™, Stronghold® (Pfizer)	Monthly topical spot-on for dogs and cats: • Fleas, adult & larvae • Heartworm prevention • Ear mite treatment • Ticks • Sarcoptic mange treatment • Hookworms • Roundworms	• Dogs & cats: 6 mg/kg topical, once per month for preventative use. • For sarcoptic mange: once every two weeks until 3 consecutive skin scrapings done one week apart are determined negative. • Safe for ivermectin-sensitive dogs
Sentinel (Novartis) Milbemycin oxime + lufenuron	• Monthly heartworm prevention + flea control (flea eggs)	• Dogs, cats: 0.5 mg/kg + 10 mg/kg PO, once per month
Spinosad	• Monthly flea prevention for dogs. • Kills adult fleas only.	• See Comfortis, Trifexis
Stronghold® Pfizer	• Monthly topical spot-on for dogs and cats against ectoparasites & endoparasites and heartworm prevention.	• See selamectin
Sulfamethoxazole + Trimethoprim	• Antiprotozoal & antibacterial • *Toxoplasma, Neospora, Hepatozoon*, coccidia	• *Toxoplasma & Neospora*, dogs & cats: 15 mg/kg PO BID x 28 days • Coccidia: 30 mg/kg PO SID x 10 days • For *Hepatozoon* need adjunct therapy with pyrimethamine & clindamycin

Drug or Product	Application	Dose & comments
Thiabendazole	• For dogs, treatment of roundworms, strongyles, lungworm	• Dogs: roundworms, strongyles: 50-60 mg/kg PO. Repeat in 3 weeks • Dogs: *Filaroides*: 35 mg/kg PO BID x 5 days, then 70 mg/kg BID x 21 days. Adjunct treatment with prednisolone and other supportive drugs may be necessary.
Trifexis (Eli Lilly) Milbemycin oxime + Spinosad	Monthly treatment for dogs: • Heartworm prevention • Adult fleas only • Hookworms • Roundworms • Whipworms	• Dogs, oral dosing once per month. • Kills only adult fleas, therefore not effective in management of flea allergy patients. • Does not kill ticks
Tri-Force (Bayer) Cyphenotherin + Nylar insect growth regulator	Monthly topical spot on: • Fleas: adult and larvae • Ticks • Mosquito repellent	• Once per month topical spot-on • Separate formulations for dogs and cats

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Appendix 5: Anesthetic and analgesic drug protocols

Abbreviations

TZ: Tiletamine 250mg / zolazepam 250mg (Telazol® in N. America; Zoletil-100® in Europe & Australia) **IM**: intramuscular **IV**: intravenous **SQ**: subcutaneous **q**: every (e.g., q 8 hrs = once every 8 hours) **CNS**: Central Nervous System

Fundamentals

For anesthetic considerations and protocols for early-age neutering, refer to Section 8: Early-Age Sterilization (EAS).

For critical considerations in anesthetic monitoring, stabilization of physiological systems during anesthesia and pre- and post-anesthetic care, refer to Section 6: Anesthesia, Appendix 7: Vital parameters during anesthesia, and Appendix 8: Guide for monitoring depth of anesthesia.

All emergency drugs and protocols must be stocked and immediately available, and staff must be trained how to use them appropriately. Refer to Appendix 9: Emergency drugs quick reference drug chart; Appendix 10: Emergency treatment kits; Appendix 11: Emergency treatment of cardiac and respiratory arrest; Appendix 12: Emergency treatment for anaphylactic reaction.

Prior to anesthesia, the animal must be as calm as possible. Stress significantly compromises the effectiveness of anesthetic drugs, and can make anesthesia unstable, necessitate the administration of much more drug, and consequently prolong recovery times.

After injection with pre-medication drugs, the patient's environment must be very quiet, and the patient must be checked at least every 5 minutes. Care must be taken to prevent arousal of the animal while s/he is going to sleep (see notes on Plane 2 of anesthesia, Appendix 8: Guide for monitoring depth of anesthesia.)

Once the drugs begin to take effect, the patient must not be left alone. The patient may vomit and not be able to protect his or her airway, convulse, or fall into a position that compromises respiration. Similarly, animals must be monitored closely throughout recovery to ensure safety and to manage emergencies immediately. Refer to Section 6.6: Anesthetic monitoring for details.

All animals must be intubated with an appropriately-sized endotracheal tube immediately on induction.

Periodic assisted ventilation throughout anesthesia is advisable (ca. every 3-5 minutes) to prevent hypercarbia (elevated CO_2 levels). If the animal is not connected to an anesthetic machine, ventilation may be performed with an ambu bag.

A variety of anesthetic combinations are used successfully in field spay / neuter projects around the world. IFAW does not promote one protocol over others: the decision must be made by the veterinarian in charge of the project on the basis of his or her experience and the availability of drugs and equipment at the project site. It is understood that ideal drugs and equipment often are not available, and that in field surgeries, risks must be accepted that are inherent in such work.

Regardless of the drug combination that is chosen, the veterinary team must do all that it can to minimize risk to the patients, and to weigh risk against benefit to individual animals and to the project. Close monitoring of patients and excellent preparation and training for emergency intervention are essential.

Anesthetic drug protocols

All anesthetic drug protocols for sterilization surgery must ensure:

1. Safety, given age and physiological predisposition of the individual patient.

   Close monitoring and preparation with emergency drugs and well-trained emergency intervention protocols are essential, and can make even non-ideal conditions and resources work well. Anesthetic monitoring procedures are detailed in Section 6.6: Anesthetic monitoring.
2. Full loss of consciousness
3. Visceral analgesia
4. Muscle relaxation
5. Surgical plane (Stage 3) anesthesia throughout the surgical procedure (cf. Appendix 8: Guide for monitoring depth of anesthesia)
6. Post-operative analgesia for a minimum of 24 hours

Anesthetic drug protocols include the following five stages:

1. Premedication: neuroleptanalgesia

- Premedication should sedate and relax the animal, and provide analgesia. Less anesthetic is subsequently required for induction and maintenance.
- Neuroleptanalgesia is produced by a combination of a sedative or tranquilizer with an analgesic. It induces a state of CNS depression (hypnosis) and analgesia. It is useful for short surgical or other painful procedures or as premedication for full anesthesia.
- Examples of common premedication neuroleptanalgesic combinations:
  - Benzodiazepine + opiate (dogs & cats)
  - Acepromazine + opiate (dogs)
  - Alpha-2 agonist + opiate ± ketamine (dogs & cats)
- Side effects, depending on drug combination: bradycardia, respiratory depression, ataxia.
- Anticholinergics (atropine, glycopyrrolate) are appropriate for use with opiates, but not necessarily with alpha-2 adrenergic drugs. See discussion below.
- Acepromazine by itself is not considered a good premedication. It is hypotensive, and causes drowsiness, but is not anxiolytic and not analgesic. Not appropriate for pediatrics. See notes below.
- Wait 5-15 minutes until the animal is relaxed and drowsy before administering the induction agent.
- The animal must be as calm as possible before pre-medication drugs are administered. The more stressed and anxious the patient is, the less effective pre-medication and subsequent anesthetic agents will be.
- It is important that the animal is kept quiet and un-stimulated during the pre-medication period.
- In some instances, the premedication step is not feasible, e.g., with highly fractious animals. In such cases, animals may be induced directly with, for example, an injection of TZ or ketamine/diazepam mixed with an opiate or alpha-2 adrenergic drug.

  (Note that dexmedetomidine cannot be mixed in the same syringe with butorphanol.)

2. Induction

- Injectable agents (e.g., Propofol, ketamine/diazepam, TZ) or an inhalant (e.g., isoflurane). Isofluorane works well for young animals undergoing Early-Age Sterilization.
- Patient is induced to Stage 3 (surgical) anesthesia and intubated.
- Topical lidocaine around the glottis and epiglottis aids in intubation and reduces the likelihood of damage due to laryngospasm. This step is essential for cats, and advisable for dogs.

3. Maintenance

- Injectable anesthetic may be used for short procedures or if inhalant anesthesia is not possible. Options for injectable anesthetics that are appropriate for use in maintenance anesthesia include pentobarbital, ketamine/diazepam, and TZ.
- Inhalant anesthetics are advised for longer procedures or for animals who may be a greater anesthetic risk or who need rapid recovery times, e.g., puppies and kittens undergoing Early-Age Sterilization, or lactating queens/bitches.
- Note that TZ or ketamine/diazepam must not be used alone for sterilization surgery. These combinations do not provide sufficient muscle relaxation or analgesia. Animals should be premedicated with a neuroleptanalgesic combination.
- Some protocols combine TZ with an alpha-2 adrenergic drug. Close monitoring is essential to prevent problems due to respiratory depression and bradycardia. This is explained in further detail below.
- The use of line blocks with lidocaine or bupivicane when closing the abdomen, as a testicular block or when closing the incision following castration may reduce the need for anesthetic top-up. Line blocks are not effective for prevention of pain of ovarian ligament tug during ovariohysterectomy.

4. Recovery

- Recovery of animals from anesthesia must be closely monitored, and animals must not be left alone until they are able to stand and walk on their own.
- Reversal drugs may be used to hasten recovery. Atipamezole is used to reverse effects of alpha-2 agonists such as xylazine, medetomidine and dexmedetomidine. This is often desirable, particularly if the patient experiences bradycardia or compromised respiration.
- Opiate antagonists (e.g., naloxone) are used to reverse the effects of opiates. Provided that the animal is in no danger, it may be preferable to let the opiate effects last as long as possible for the post-operative analgesic and relaxing effects of the drug.
- When reversing drugs, be sure to protect staff and the patient from the excitatory effects that may emerge with rapid arousal. Be aware also that reversal of a drug will remove not only the sedative effects, but also the analgesic effects that the drug had provided (e.g., with opiates).
- Analgesia must be in effect before the animal wakes up, and continue for at least 24 hours post-operatively.

5. Post-operative analgesia

- Usually achieved with non-steroidal anti-inflammatory drugs, tramadol or long-acting opiates (e.g., buprenorphine).
- All animals subjected to sterilization surgery must receive analgesic medication that lasts for at least 24 hours after surgery.
- Analgesia must be in effect before the animal recovers from anesthesia. Do not allow an animal to wake up in pain and then try to subdue pain. In addition to being inhumane, this results in the need for a much higher dose of drug.
- Further discussion of post-operative analgesia are described in the next section, as well as in Appendix 6: Assessing the need for post-operative analgesia.

6. Post-operative analgesia

Analgesia must be provided to all animals subjected to surgery and should last for at least 24 hours after sterilization surgery. Some individuals may require longer periods of analgesia if they are experiencing inflammation or pain at the surgical site.

It is important that analgesics are administered before pain begins. Preventing the development of pain is more effective, more cost-effective and more humane than at trying to stop pain once has begun. Lower doses are required to control pain pre-emptively.

To control post-operative pain, non-steroidal anti-inflammatory drugs (NSAIDs) or tramadol may be given anytime during surgery, before the patient wakes up. Opiates should be given prior to surgery and continued, as required, during and after surgery. Opiates are often used in the premedication mixture (see above).

NSAIDs and tramadol provide good post-operative pain relief, but are not suitable for intraoperative analgesia. Intra-operative analgesia requires opiates or alpha-2 adrenergic drugs.

For postoperative analgesia in healthy, non-pediatric, dog and cat spays and neuters, a preferential COX-2 inhibitor NSAID may be used. Analgesia usually lasts for ca. 24 hours, and further doses are usually not required. The need for further analgesia should be evaluated on a case-by-case basis (Appendix 6: Assessing the need for post-operative analgesia).

In Early-Age Sterilization patients, opiates are preferred over NSAIDs for post-operative analgesia. Buprenorphine is often a good choice.

Individuals who need longer periods of post-operative analgesia should be evaluated to ensure that the patient is in good health and that further treatment with an NSAID will not be detrimental to his or her health. Buprenorphine is the analgesic of choice for cats. Healthy cats usually tolerate meloxicam well for 3-4 days. An opiate or NSAID, or a combination of the two, may be used for dogs.

Glucocorticoids must never be administered perioperatively (before, during or after surgery).

Glucocorticoids must never be administered together with NSAIDs.

Opiate doses for intra-operative and post-operative analgesia: see comments below under “Opiates”

Tramadol for post-operative pain: see below under “Tramadol”

Non-steroidal anti-inflammatory drugs (NSAID)

- NSAIDs are the most commonly used drugs to control post-operative pain after sterilization surgery.
- Whenever possible, use preferential COX-2 inhibitors (e.g., meloxicam, carprofen, etodolac, deracoxib). NSAIDs with selective or equivalent COX-1 activity are associated with higher gastrointestinal side effects (ulceration, vomiting, anorexia).
- Cyclooxygenase inhibitors with anti-thromboxane activity (e.g., aspirin, tolfenamic acid) should be avoided perioperatively because of potential bleeding risk.
- NSAIDs should not be given to sterilization surgery patients with the following conditions. Use an opiate instead to control pain in these patients.
    - Dehydration, hypovolemia, hypotension
    - Where surgical complications have resulted in cardiovascular or renal compromise.
    - If the animal received any exogenous corticosteroids recently. There must be at least one week interval between the last dose of short/intermediate duration glucocorticoids (prednisone, prednisolone, and triamcinolone) and the start of an NSAID. This interval is called a ‘washout’ period. For long-acting glucocorticoids such as dexamethasone and betamethasone, the washout period must be 3-4 weeks before starting an NSAID.
- Switching from one NSAID to another may be done only with an appropriate wash-out period between drugs. If the animal has been receiving an NSAID, and the veterinarian finds it necessary to change to a different NSAID, there must be a washout period of 3-7 days between the cessation of the first NSAID and beginning the new one. If the first NSAID was aspirin, the washout period must be two weeks before the new NSAID is started.
- The advantages of using NSAIDs instead of opioids for controlling post-surgical pain include:
    - Dose needs to be given only once per day
    - Less stringent drug control laws than with opioids, and therefore more readily available in some areas.
    - No sedative or dysphoric effects as seen sometimes with opioids. These effects may interfere with the assessment of pain and the determination of whether pain is properly controlled in the patient.
- Carprofen appears to have COX-2 specificity in dogs; less so in cats. It is highly bound to plasma proteins and has a low volume of distribution, with a primarily hepatic metabolism. Therefore, use with care in young animals, in animals with low plasma protein levels, or in animals with compromised liver function (one wouldn’t be performing sterilization surgery on the latter two anyway). May be used in cats as a single dose for control of post-operative pain if meloxicam is not available.
    - Dogs: 2-4 mg/kg SQ. May repeat 24 hrs later with oral administration at 2 mg/kg for up to 5 days.
    - Cats: 2-4 mg/kg, SQ.

- Meloxicam is also a COX-2 preferential NSAID, and is usually well tolerated by cats and dogs.
  - Dogs: 0.2 mg/kg SQ at time of surgery. May continue with 0.1 mg/kg PO (oral suspension, 1.5 mg/ml) q 24 hours for 3-5 days thereafter if needed.
  - Cats: 0.2 mg/kg SQ at time of surgery. If subsequent doses are necessary, administer the second dose at 0.1 mg/kg SQ or PO 24 hours later, then 0.025 mg/kg q 24-48 hrs.
- Tepoxalin (Zubrin®).
  - Unclear whether preferential COX-2 inhibitor.
  - Available only as an oral compound, so not suitable for injectable administration while the animal is anesthetized.
  - May be used as an oral NSAID in dogs requiring pain control beyond 24 hours after surgery. However, if dogs were given an injection of another NSAID prior to waking up from anesthesia, they should be continued on the same drug if further doses of an NSAID are needed.
  - Dog: first dose 20 mg/kg PO; subsequent doses should be 10 mg/kg SID if needed.
  - Not approved for use in cats.
- Deracoxib (Deramaxx®)
  - Selective COX-2 inhibitor. Available only as an oral compound, so not suitable for injectable administration while the animal is anesthetized.
  - May be used as an oral NSAID in dogs requiring pain control beyond 24 hours after surgery. However, if dogs were given an injection of another NSAID prior to waking up from anesthesia, they should be continued on the same drug if further doses of an NSAID are needed.
  - A related drug, fibrocoxib (Previcox®) is recommended for management of osteoarthritic pain only. Deracoxib is recommended for management of post-operative pain as well as for osteoarthritic pain.
  - Dogs: 3-4 mg/kg once per day, maximum 7 days. This is the dose for post-operative pain management. The long-term osteoarthritis pain management dose is half of this.
  - Deracoxib has not been sufficiently tested in cats to recommend use in this species.
- Ketoprofen (Ketofen®, Kepplin® and other trade names)
  - Ketoprofen is COX-1 selective, and therefore not suitable for repeated dosing. It is available as injectable and oral forms.
  - Ketoprofen may be used if an appropriate COX-2 selective NSAID or opiates are not an option.
  - Dogs: 2 mg/kg SQ at the time of surgery. If analgesia is necessary beyond 24 hours, dosing with oral ketoprofen at 1 mg/kg may be repeated once per 24 hours for no more than 5 days total.
  - Cats: 2 mg/kg SQ at the time of surgery. If analgesia is necessary beyond 24 hours, dosing with oral ketoprofen at 1 mg/kg may be repeated once per 24 hours for no more than 3 days total.
- Etodolac (Etogesic®, Lodine® and other trade names)
  - Selective COX-2 inhibitor. Available only as an oral compound, so not suitable for injectable administration while the animal is anesthetized.
  - May be used as an oral NSAID in dogs requiring pain control beyond 24 hours after surgery. However, if dogs were given an injection of another NSAID prior to waking up from anesthesia, they should be continued on the same drug if further doses of an NSAID are needed.
  - Dose for dogs: 10-15 mg/kg once per day.
  - Etodolac has not been sufficiently tested in cats to recommend use in this species

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NSAID Recommendations for Postoperative Pain Control in Dogs (2 to 3 days' duration)[1]

Drug	Formulations	Dose[2]	Frequency
Carprofen	Oral tablets and injectable	4.4 mg/kg (first dose), thereafter 2 mg/kg	q 24 hr
Meloxicam	Oral suspension and injectable	0.2 mg/kg (first dose), thereafter 0.1 mg/kg	q 24 hr
Ketoprofen	Injectable	2.0 mg/kg (first dose), thereafter 1 mg/kg (IV, IM)	q 24 hr up to 5 days (dog) or 3 days (cat)
Deracoxib	Oral tablets	3.0-4.0 mg/kg	q 24 hr up to 7 days
Etodolac	Oral tablets	10.0-15.0 mg/kg	q 24 h

1: All dose recommendations are based on manufacturer guidelines for oral dosing except where otherwise indicated.

2: Following the initial dose, assessment of analgesic efficacy should be continued and subsequent dosing adjusted to achieve the minimum effective dose.

Table adapted from: Tranquili, W.J. and Grimm, K.A. 2003. Pain management alternatives for common surgeries. Pain Management Symposium, North American Veterinary Conference and the American Animal Hospital Association Annual Meeting, Orlando, FL.

Notes on specific drugs and drug combinations

Acepromazine (phenothiazine)

- Acepromazine produces sedation and muscle relaxation, but does not relieve anxiety. If patients are stressed or anxious, acepromazine may make them feel even worse, as they no longer have good control over their muscles but are still anxious. (Don't use it for transporting dogs and cats.) The sedative effect of acepromazine can be overwhelmed with stimuli and anxiety. It is important to keep the animal in a quiet, darkened place with minimal activity during sedation.
- Antiemetic.
- No analgesic effect, though may potentiate analgesic effects of other drugs.
- Do not use alone as a premedication agent. See list above for appropriate premedication options.
- Hypotensive, particularly in apprehensive or excited animals; usually results in tachycardia as a result.
- Metabolized by the liver: avoid use in pediatric patients and in patients with compromised hepatic function.
- Lowers seizure thresholds on animals with epilepsy, but may inhibit chemically-induced seizures, e.g., from ketamine.
- Phenothiazine tranquilizers should not be used in animals younger than 12 weeks of age. It may be administered at 0.025-0.05 mg/kg subcutaneously in animals 12-16 weeks of age. In immature animals, phenothiazines produce prolonged CNS depression and potentiation of hypotension and hypothermia.

Alpha 2 agonists (xylazine, medetomidine, dexmedetomidine)

- Pronounced sedation. Good muscle relaxation. Some analgesia.
- Minor analgesia when used alone. Enhance the potency and duration of analgesia of opioids given in pre-anesthetic mixtures (neuroleptanalgesia).
- Cause vomiting in dogs & cats (especially in cats). Reduce salivation, gastrointestinal motility and swallowing reflex.
- Respiratory depression
- Bradycardia and decreased cardiac contractility (decreased cardiac output and blood pressure) secondary to increased peripheral vascular resistance. May last longer with dexmedetomidine than with xylazine. Be careful with anticholinergics – see next point.
- The use of atropine or glycopyrrolate is controversial with alpha-2 agonists. The decreased blood pressure caused by alpha-2 agonists results in a reflexive increase in peripheral vascular resistance. The heart has to pump against this resistance, and the mechanism to protect the heart in response to this increase in blood pressure is to slow the heart rate (compensatory bradycardia). Atropine works against this compensatory bradycardia, forces the heart rate to stay high in the face of increased peripheral resistance, and thereby increases the risk of myocardial ischemia.
- In the clinical setting, the bradycardia induced by alpha-2 agonists is monitored closely, together with blood pressure and pulse. Atropine is given if bradycardia is marked and compromises blood pressure. Atipamezole to reverse the alpha-2 agonist activity may be a better choice of drug with cardiovascular compromise (see below).
- Peripheral vasoconstriction combined with respiratory depression results in pale, purplish or gray mucous membranes. This is concerning, but be aware that it is an expected consequence of alpha-2 agonist activity and monitor cardiovascular status closely.
- Periodic assisted ventilation throughout anesthesia is advisable (ca. every 5 minutes) to prevent hypercarbia (elevated CO_2 levels).
- Hypothermia
- Metabolized by the liver: avoid use in pediatric patients and in patients with compromised hepatic function.
- Dexmedetomidine is not licensed for use in the lactating bitch or queen; it is transmitted in the milk in humans. It may have an adverse effect on the offspring.
- Avoid use of alpha-2 agonists in pediatric patients (cardiovascular effects & hepatic metabolism – cf.

Section 8: Early-Age Sterilization (EAS).

- Dexmedetomidine dose for premedication:
  - Dog: 0.125-0.375 mg/m^2
  - Note that the canine dosage for dexmedetomidine is given in mg/m^2 body surface area rather than mg/kg body weight. A conversion chart is provided at the end of this appendix.
  - Cat: 0.01-0.04 mg/kg
  - In combination with an opioid, use 0.005 mg/kg (5 mcg/kg) for dogs & cats.
  - Dexmedetomidine cannot be mixed in the same syringe with butorphanol.
- Xylazine dose for premedication:
  - Dog: 0.5-1.5 mg/kg
  - Cat: 0.5-1.5 mg/kg
- Reverse with yohimbine or atipamezole – see section on Drug Reversal, below.
- Accidental human exposure
- Dexedetomidine, medetomidine and atipamezole can be absorbed through skin abrasions and mucous membranes. As little as 0.1 ml of dexmedetomidine can cause hypotension and sedation in humans. If dexmedetomidine or atipamezole is spilled onto human skin or mucous membranes, it must be washed off immediately. If accidental exposure or injection occurs, medical attention should be sought immediately.

Barbiturates: Pentobarbital (Nembutal®), Thiopental (Pentothal®)

- Pentobarbital is a short-acting barbiturate (20-30 minutes); thiopental is ultra-short acting (5-15 minutes). Used for short procedures, or for induction of anesthesia, followed by maintenance with inhalant anesthesia.
- Note that pentobarbital is now outdated as an anesthetic. Its principal use today is as an ingredient in euthanasia solutions. It may still be used as an anesthetic in some developing areas of the world.
- Barbiturate drugs are good sedatives but do not have intrinsic analgesic activity. If performing surgery, it is essential to use a maintenance anesthetic with analgesic properties.
- Barbiturates do not produce good muscle relaxation on their own. Maintenance anesthesia should contain a good muscle relaxant, as well as analgesic drug.
- Premedicate with atropine or glycopyrrolate to decrease salivary secretions, laryngospasm and vagal activity (hypotension).
- Dose-dependent respiratory and cardiovascular depression. Apnea is most common with rapid IV administration. If a patient is already in a surgical plane of anesthesia, small additional doses of barbiturate may cause a profound drop in blood pressure. Top up with care and monitor patient closely.
- Cardiovascular effects: hypotension, tachycardia, reduced stroke volume.
- Perivascular injection is extremely painful and causes tissue necrosis. An intravenous catheter should be placed for injection of barbiturates.
- Periodic assisted ventilation throughout anesthesia is advisable (ca. every 3-5 minutes) to prevent hypercarbia (elevated CO_2 levels).
- Not to be used for Caesarean sections, as barbiturates readily cross the placenta and cause fetal respiratory arrest at doses that do not produce anesthesia in the mother.
- Patients are usually depressed for several hours after recovery from barbiturate anesthesia (even with an ultra-short acting barbiturate), depending on the dose. Repeated doses are cumulative.
- Barbiturates have a small margin of safety. Pentobarbital dose varies from 10-30 mg/kg; minimum lethal dose in dogs is 50 mg/kg. For euthanasia, use at a minimum of 150 mg/kg.
- Pentobarbital depends on hepatic metabolism for clearance. Do not use in pediatric patients or for patients with compromised liver function. Repeated doses are cumulative.

- Thiopental: use at 6-12 mg/kg IV for induction of anesthesia. Do not use at concentrations greater than 10% solution, as these cause severe tissue damage.
- Thiopental (1.25% solution) may be used with great care in puppies and kittens older than 12 weeks. For pediatric patients, it is preferable to use thiopental only as an induction agent, followed by an inhalant anesthetic for maintenance.
- Barbiturates are the principal component of euthanasia solutions, as they induce cardiac and respiratory arrest at high doses.

Benzodiazepines (diazepam, midazolam)

- Muscle relaxation, anticonvulsant. Benzodiazepines are not used for analgesia.
- Little cardiopulmonary effects unless given rapidly IV.
- Benzodiazepines result in minimal sedation when used alone. In cats, may see paradoxical anxiety leading to aggression. However, when added to an opiate, benzodiazepines appear to produce a synergistic sedative effect (neuroleptanalgesia) and, when added to ketamine, will improve muscle relaxation.
- Absorption of diazepam is slower than that of midazolam, so onset of action will be slower. Diazepam should be given only IV or orally. IM injection of diazepam is painful due to the propylene glycol carrier, and is absorbed slowly from the injection site. Midazolam is water soluble and appropriate for IM injection.

Dissociative anesthetics: ketamine and tiletamine (tiletamine is always in combination with zolazepam-Telazol® or Zoletil®)

- Dissociative anesthetics interrupt communication between areas of the brain that control conscious and unconscious activity. The resulting cataleptic state is characterized by a loss of appropriate response to external stimuli, muscle rigidity, and loss of muscle control.
- Used for restraint, induction of anesthesia, and minor, short procedures that do not require visceral analgesia.
- Dissociative anesthetics produce some superficial analgesia, but do not provide visceral analgesia, e.g., for spay and neuter surgery.
- When used for sterilization surgeries, dissociatives must be combined with neuroleptanalgesic premedication. Alone, ketamine and TZ do not produce sufficient muscle relaxation and do not produce sufficient visceral analgesia.
- Oral, ocular and swallowing reflexes remain intact with ketamine. Eyes remain open; eye position usually is rotated.
- It is important to instill an ocular lubricant (artificial tear ointment) into the eyes during anesthesia with dissociative anesthetics to protect corneas.
- Ketamine must be combined with a sedative or benzodiazepine to counteract muscle rigidity. TZ, although containing zolazepam, also does not provide good muscle relaxation and should be combined with an additional drug (e.g., dexmedetomidine). See section above on premedication.
- Seizures are common with ketamine, particularly in high doses. (Some species other than domestic cats and dogs are prone to seizures with ketamine even at lower doses.) “Ketamine shakes” occur in ca. 30% of dogs and cats at induction if ketamine is not used in combination with a benzodiazepine or opiate.
- Apneustic breathing (long inhalation phase), particularly on IV induction with ketamine. Monitor closely to ensure sufficient oxygenation, and that the breathing pattern returns to normal within a few minutes. As always, ensure that the animal is intubated immediately following induction.
- Periodic assisted ventilation throughout anesthesia is advisable (ca. every 5 minutes) to prevent hypercarbia (elevated CO_2 levels).
- Cardiovascular effects: heart rate and blood pressure increase with concomitant decrease in cardiac contractility. Contraindicated in animals with pre-existing heart disease. Take care with anticholinergics (see next point).

- The routine use of anticholinergics as a pre-med with TZ is controversial. Anesthesiologists recommend that atropine be reserved for treatment of bradycardia when it occurs. See comment above with regard to alpha-2 agonists.
- Animals are ataxic and hyper-responsive during recovery from dissociative anesthesia. It is critical to maintain a quiet, dark environment during recovery. Animals must be confined in a manner that prevents self-injury during recovery, and must be monitored closely to provide immediate intervention if necessary.
- There is no reversal drug for dissociative anesthetics.
- Metabolized by the liver, excreted via kidneys. Not advisable for use in animals less than 12 weeks of age, or in animals with compromised hepatic or renal function.
- Dogs tend to have more problems with TZ than cats: poor muscle relaxation, respiratory depression and ‘hangover’ post-recovery. Keep dose to a minimum by combining with appropriate other drugs and minimizing repeated doses.
- Generally, the IV dose for TZ is 20-50% of the IM dose.

Inhalant anesthetics (isoflurane, sevoflurane)

- Advantages of gas anesthetics over injectable anesthetics:
  - Depth of anesthesia is more quickly and easily regulated, hence safer to use
  - Induction and recovery are usually smooth and rapid
  - Oxygen is delivered simultaneously
  - Inhalant anesthetic drugs are minimally processed by liver and kidney
  - Generally fewer undesirable effects
- Periodic assisted ventilation throughout anesthesia is advisable (ca. every 3-5 minutes; more frequently for paediatric patients) to prevent hypercarbia (elevated CO_2 levels).
- Excreted primarily from lungs, little biodegradation, hence safer option than injectable anesthetics for pediatric patients and for patients with hepatic or renal compromise.
- Note that induction by masking an animal down or in a chamber is not appropriate. The animal experiences a high degree of stress during this kind of induction. The process is associated with severe sympathomimetic effects, bronchial irritation, and increased risk of aspiration of gastric contents.
- Isoflurane: excellent muscle relaxation, hypotensive effects are dose-dependent, readily crosses placenta. Induction is facilitated by premedication with a neuroleptanalgesic combination, or with an injectable induction agent (e.g., Propofol). Maintenance dose is generally 1-3%.
- Sevoflurane: blood-gas partition coefficient is ca. ½ that of isoflurane, hence more rapid induction and recovery, requiring less drug. Other than this, its properties and clinical use are similar to those of isoflurane. Maintenance dose is generally 3-4%.
- Sevoflurane should not be used with soda lime as absorption compound: produces potentially toxic product on interaction with soda lime.
- Note that each drug requires a dedicated and properly-calibrated vaporizer. Do not fill an isoflurane vaporizer with sevoflurane, or vice versa. Vaporizers must be maintained and calibrated per manufacturer’s instructions.

Local anesthetics (lidocaine, bupivicaine)

- These drugs work on sodium channels that are active in pain and inflammation. In addition to analgesia, lidocaine and bupivicaine cause vasodilation, which promotes healing and counteracts inflammation.
- Bupivicaine has a longer duration to onset of action than lidocaine (20-30 min vs. 5-10 min), but has a longer duration of effect (6-8 hours vs. 1-2 hours). Mixing the 2 together will result in a longer time to onset of effect than when using lidocaine alone, and a shorter duration of action than when using bupivicaine alone, thereby diluting the desirable properties of each drug.

- Care must be taken to avoid exceeding maximum dose, beyond which toxicity (arrhythmias) may occur. Bupivicaine maximum dose is 1-2 mg/kg per 8 hours. Lidocaine maximum dose for dogs is 4 mg/kg; for cats 2-3 mg/kg.
- Often beneficial to use as line block at closing the abdomen, as a testicular block, or when closing the incision at castration. Will not provide analgesia for ovarian tug in spays. Mix lidocaine or bupivicaine with saline, squirt into incision and close over it (don't need to inject into tissue).
- Do not inject bupivacaine IV: may precipitate severe cardiac arrhythmias that are not responsive to treatment. Lidocaine should be injected IV only if intentionally used to stop ventricular arrhythmias (in which case the patient should be connected to an ECG machine).

Opiates (butorphanol, buprenorphine, morphine, oxymorphone)

- Opiates produce analgesia and sedation without loss of proprioception or consciousness. Different types of opiates have varying affinities for different opiate receptors (mu, kappa, delta, sigma). Some are full agonists (morphine), others partial agonists (buprenorphine), or agonists/antagonists (butorphanol), with associated advantages and disadvantages.
- Opiates may induce paradoxical excitatory effects, especially full agonists (morphine, oxymorphone), and especially in cats. If given rapidly IV, may induce excitation in dogs as well.
- Use in a neuroleptanalgesia cocktail (opiate combined with acepromazine, benzodiazepine or alpha-2 agonist) to avoid excitatory or dysphoric effects, particularly in cats.
- Butorphanol and buprenorphine have fewer excitatory side effects than morphine, are less sedating than morphine, and have better analgesic properties (buprenorphine 25-30 times; butorphanol 4-7 times)
- Cardiovascular effect: depending on the drug and dose, may see bradycardia, hypotension. Include atropine or glycopyrrolate in premedication cocktail.
- Respiratory depression (rate & tidal volume) is dose-dependent. Normally not seen at low doses unless patient is already depressed or unconscious.
- Periodic assisted ventilation throughout anesthesia is advisable (ca. every 3-5 minutes) to prevent hypercarbia (elevated CO_2 levels).
- Body temperature may decrease in dogs and increase in cats.
- Gastrointestinal effects: salivation, vomiting, ileus.
- Combined with tranquilizers and sedatives, opiates produce neuroleptanalgesia. Use as a premedication or with anesthetics to produce balanced anesthesia – see notes on premedication, above.
- Butorphanol is rapid-onset, short acting (2-4 hrs), while buprenorphine has slower onset but longer duration of action (4-12 hrs). It is possible to give butorphanol as premedication and buprenorphine for post-operative analgesia. When using both drugs, deliver them at least 45 minutes apart.
- Do not mix butorphanol and buprenorphine together. The result would be a combination that has a slower onset of action than butorphanol and a shorter duration of action than buprenorphine, thereby diluting the desirable properties of each drug.

Parenteral Opioid Recommendations for Perioperative Pain Management

Drug	Canine Dose[1]	Feline Dose[1]
Butorphanol	0.2-0.4 mg/kg q 1-2 hr SQ IM lower dose IV	0.1-0.4 mg/kg q 1-2 hr SQ IM lower dose IV
Buprenorphine[2]	0.005-0.02 mg/kg q 4-12 hr IM SQ IV	0.005-0.01 mg/kg q 4-12 hr IM SQ IV May be squirted into open mouth at 0.02 mg/kg in fractious cats.
Fentanyl	5.0-10.0 μg/kg/hr IV or CRI	2.5-5.0 μg/kg/hr IV or CRI
Hydromorphone	0.05-0.2 mg/kg q 2-6 hr IM SQ IV	0.05-0.1 mg/kg q 2-6 hr IM SQ IV
Methadone	0.1-0.5 mg/kg q 4-6 hr IM SQ	0.05-0.5 mg/kg q 4-6 hr IM SQ
Morphine	0.5-1.0 mg/kg q 2-6 hr IM SQ	0.1-0.3 mg/kg q 4-8 hr IM SQ
Oxymorphone	0.05-0.2 mg/kg q 2-4 hr IM SQ	0.05-0.1 mg/kg q 2-6 hr IM SQ

IM = intramuscular   SQ = subcutaneous   IV = intravenous   CRI = constant rate infusion

1: Doses should be halved for pediatric patients. Note pediatric considerations for opiates (cf. Section 8: Early-Age Sterilization (EAS))

2: Higher doses of buprenorphine (0.02-0.03 mg/kg) may achieve a longer duration (10 hours) of effect, but will not necessarily improve analgesia.

Table adapted from: Tranquili, W.J. and Grimm, K.A. 2003. Pain management alternatives for common surgeries. Pain Management Symposium, North American Veterinary Conference and the American Animal Hospital Association Annual Meeting, Orlando, FL.

Propofol

- Rapid-acting, ultra-short duration, non-barbiturate anesthetic. Produces sedation-hypnosis similarly to thiopental.
- Good muscle relaxation, rapid recovery with little 'hangover effect'
- Dose-dependent respiratory depression and hypotension.
- Poor analgesia.
- Generally used as induction agent prior to maintenance with gas anesthesia. If used for general anesthesia for short procedures (e.g., Caesarean section), must combine with opioid analgesics or other neuroleptanalgesic combinations.
- Delivered intravenously (despite milky color). Some pain on initial injection.
- Biotransformed by liver; excretion hepatic and extrahepatic. Repeated doses are relatively non-cumulative compared with barbiturates.
- Propofol is the induction agent of choice for young patients. However, animals with immature hepatic function should not receive repeated doses, as this may prolong recovery.
- Expensive.
- Dose: 3-6 mg/kg IV. Deliver 25% of the calculated dose every 30 seconds until the desired effect is achieved. Premedication with tranquilizer or sedative will reduce the dose needed for induction.

Tramadol

- Analgesic drug. Effective for management of post-operative pain. Not effective for control of pain during surgery.
- Synthetic mu-receptor opiate agonist that also inhibits reuptake of serotonin and norepinephrine
- Appears to be well tolerated in dogs. Be careful with use in cats due to feline hypersensitivity to opiates. Cats may suffer narcotizing effect with tramadol.
- Dogs & cats: 2-4 mg/kg SQ, IM or IV. Oral tablets are also available (2-4 mg/kg PO q8-12 hours)

Drug reversal

- Alpha-2 adrenergic drugs may be reversed with atipamezole if a rapid recovery is desirable, e.g., if the animal is experiencing problems with anesthesia, or if recovery from anesthesia is prolonged.
- Yohimbine for reversal of xylazine: 0.11 mg/kg slow IV (dogs)
- Atipamezole (Antisedan®): reversal of medetomidine, dexmedetomidine and xylazine
    - Atipamezole to reverse dexmedetomidine:
        - Note that dosage for atipamezole to reverse dexmedetomidine is given in mg/m^2 body surface area. See conversion chart at the end of this appendix.
        - If dexmedetomidine was given IV, give atipamezole:

          Dog: 3.75 mg/m^2 IM

          Cat: 1.875 mg/m^2 IM
        - If dexmedetomidine was given IM, give atipamezole:

          Dog: 5.0 mg/m^2 IM

          Cat: 2.5 mg/m^2 IM
    - Atipamezole to reverse medetomidine:
        - Dogs: use 5 mg (1 ml) atipamezole for each 1 mg (1 ml) medetomidine that was used.
        - Cats: use 2.5 mg (0.5 ml) atipamezole for each 1 mg (1 ml) medetomidine that was used.
        - Give IM or carefully IV. If it has been more than 45 minutes since medetomidine was delivered, give ½ the dose of atipamezole.
    - Atipamezole to reverse xylazine: Start with 0.05 mg/kg atipamezole IM. If no response or insufficient response after 10-15 minutes, repeat. Maximum dose 0.2 mg/kg (= 4 doses).
- Effects of opiate agonists (morphine, oxymorphone) may be reversed with opioid antagonists, e.g., naloxone, 0.005-0.015 mg/kg IV or naltrexone, 0.05- 0.1 mg/kg SQ.

Dexmedetomidine dosing chart for premedication in dogs (0.125 – 0.375 mg/kg), based on kg body weight and m^2 body surface area. For cats use low end of dose.

		dexmedetomidine (mg)				dexmedetomidine (mg)				dexmedetomidine (mg)	
kg	m^2	0.125 mg/m^2	0.375 mg/m^2	kg	m^2	0.125 mg/m^2	0.375 mg/m^2	kg	m^2	0.125 mg/m^2	0.375 mg/m^2
0.5	0.06	0.01	0.02	16	0.64	0.08	0.24	32	1.02	0.13	0.38
1	0.10	0.01	0.04	17	0.67	0.08	0.25	33	1.04	0.13	0.39
2	0.16	0.02	0.06	18	0.69	0.09	0.26	34	1.06	0.13	0.40
3	0.21	0.03	0.08	19	0.72	0.09	0.27	35	1.08	0.14	0.41
4	0.25	0.03	0.10	20	0.74	0.09	0.28	36	1.10	0.14	0.41
5	0.30	0.04	0.11	21	0.77	0.10	0.29	37	1.12	0.14	0.42
6	0.33	0.04	0.13	22	0.79	0.10	0.30	38	1.14	0.14	0.43
7	0.37	0.05	0.14	23	0.82	0.10	0.31	39	1.16	0.15	0.44
8	0.40	0.05	0.15	24	0.84	0.11	0.32	40	1.18	0.15	0.44
9	0.44	0.05	0.16	25	0.86	0.11	0.32	42	1.22	0.15	0.46
10	0.47	0.06	0.18	26	0.89	0.11	0.33	44	1.26	0.16	0.47
11	0.50	0.06	0.19	27	0.91	0.11	0.34	46	1.30	0.16	0.49
12	0.53	0.07	0.20	28	0.93	0.12	0.35	48	1.33	0.17	0.50
13	0.56	0.07	0.21	29	0.95	0.12	0.36	50	1.37	0.17	0.51
14	0.59	0.07	0.22	30	0.98	0.12	0.37	52	1.41	0.18	0.53
15	0.61	0.08	0.23	31	1.00	0.12	0.37	54	1.44	0.18	0.54

Equation for calculating conversion from body weight (g) to body surface area (m^2) for dogs:

$$m^2 = \frac{(10.1) \times (\text{body weight in grams})^{2/3}}{10{,}000}$$

Atipamezole dosing chart for DOGS following dexmedetomidine given IM or IV.

body weight (kg)	m^2	atipamezole, mg after IV dexmed	atipamezole, mg after IM dexmed	body weight (kg)	m^2	atipamezole, mg after IV dexmed	atipamezole, mg after IM dexmed	body weight (kg)	m^2	atipamezole, mg after IV dexmed	atipamezole, mg after IM dexmed
0.5	0.06	0.24	0.32	16	0.64	2.40	3.21	32	1.02	3.82	5.09
1	0.10	0.38	0.51	17	0.67	2.50	3.34	33	1.04	3.90	5.20
2	0.16	0.60	0.80	18	0.69	2.60	3.47	34	1.06	3.98	5.30
3	0.21	0.79	1.05	19	0.72	2.70	3.60	35	1.08	4.05	5.40
4	0.25	0.95	1.27	20	0.74	2.79	3.72	36	1.10	4.13	5.51
5	0.30	1.11	1.48	21	0.77	2.88	3.84	37	1.12	4.21	5.61
6	0.33	1.25	1.67	22	0.79	2.97	3.96	38	1.14	4.28	5.71
7	0.37	1.39	1.85	23	0.82	3.06	4.08	39	1.16	4.36	5.81
8	0.40	1.52	2.02	24	0.84	3.15	4.20	40	1.18	4.43	5.91
9	0.44	1.64	2.19	25	0.86	3.24	4.32	42	1.22	4.58	6.10
10	0.47	1.76	2.34	26	0.89	3.32	4.43	44	1.26	4.72	6.29
11	0.50	1.87	2.50	27	0.91	3.41	4.55	46	1.30	4.86	6.48
12	0.53	1.99	2.65	28	0.93	3.49	4.66	48	1.33	5.00	6.67
13	0.56	2.09	2.79	29	0.95	3.58	4.77	50	1.37	5.14	6.85
14	0.59	2.20	2.93	30	0.98	3.66	4.88	52	1.41	5.28	7.04
15	0.61	2.30	3.07	31	1.00	3.74	4.98	54	1.44	5.41	7.21

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Atipamezole dosing chart for CATS following dexmedetomidine given IM or IV.

body weight (kg)	m^2	atipamezole, mg after IV dexmed	atipamezole, mg after IM dexmed	body weight (kg)	m^2	atipamezole, mg after IV dexmed	atipamezole, mg after IM dexmed
0.5	0.06	0.12	0.16	4.25	0.27	0.50	0.66
1	0.10	0.19	0.25	4.5	0.28	0.52	0.69
1.25	0.12	0.22	0.29	4.75	0.29	0.54	0.71
1.5	0.13	0.25	0.33	5	0.30	0.55	0.74
1.75	0.15	0.28	0.37	5.25	0.31	0.57	0.76
2	0.16	0.30	0.40	5.5	0.31	0.59	0.79
2.25	0.17	0.33	0.43	5.75	0.32	0.61	0.81
2.5	0.19	0.35	0.47	6	0.33	0.63	0.83
2.75	0.20	0.37	0.50	6.25	0.34	0.64	0.86
3	0.21	0.39	0.53	6.5	0.35	0.66	0.88
3.25	0.22	0.42	0.55	6.75	0.36	0.68	0.90
3.5	0.23	0.44	0.58	7	0.37	0.69	0.92
3.75	0.24	0.46	0.61	7.25	0.38	0.71	0.95
4	0.25	0.48	0.64	7.5	0.39	0.73	0.97

Appendix 6: Assessing the need for post-operative analgesia

Surgery causes trauma to tissues. Tissue trauma results in inflammatory reactions that cause pain. By definition, therefore, surgery results in pain post-operatively. When performing any kind of surgery, it is essential to:

a) Prevent pain as much as possible;

b) Observe the patient very closely during surgery and afterwards, and treat pain that the animal experiences.

A key principle to pain management is that it is much more efficient to prevent the development of pain than to diminish pain once it has begun. Prevention of pain requires less drug and causes less stress to the animal than treatment of pain once it has begun.

Provision of medication to prevent post-operative pain is an essential element of surgery. For sterilization surgeries in dogs and cats, an injection of an NSAID, tramadol or long-acting opiate is given before the animal wakes up to prevent, or minimize, the post-operative pain that would otherwise develop.

Careful tissue handling is also essential during surgery. The more trauma a surgeon causes to tissues, the more inflammation will develop. Inflammation results in pain, and potentially compromised healing and post-surgical complications. Trauma is caused by such things as over-handling tissues, tissues dehydrating, rough handling of tissues, tearing, tugging, squeezing or otherwise stressing organs or muscle, cutting unnecessarily large incisions, cutting through muscle instead of the linea alba to enter the abdominal cavity, undermining tissue where it is not necessary, and rubbing tissue with gauze (gently blot instead). Muscle and skin sutures must place tissue edges in secure, close apposition, but not tied so tightly that tissue is strangulated. Surgeons must be properly trained, careful, prepared for the unexpected, and skilled in gentle, efficient tissue handling.

The most common cause of dogs and cats licking and chewing at their surgery site is because of skin trauma from rough clipping or shaving of fur, and not because of the surgery site itself. It is extremely important to be very gentle while shaving and preparing the animal for surgery. Rough clipping or clipping with a dirty, blunt clipper blade results in nicked skin which becomes itchy and uncomfortable. The skin nicks may not be overtly visible: just because there aren't bleeding gashes doesn't necessarily mean that the clippers didn't do harm. In cats and immature animals one must be particularly careful to avoid nicking a nipple. The skin on and around the scrotum is soft and sensitive. The animal licks at the traumatized skin, and then licks at the surgery site as well, pulls out the sutures, and now you have a problem.

Some dogs and cats return to normal behavior almost immediately after sterilization surgery, and others seem to take a bit longer to recover. Most healthy animals who undergo sterilization surgery do not require more than the single injection of 24-hour pain medication that is given at the time of surgery. Some animals need more, however, and may need pain medication for several days after surgery. Differences in age, breed, genetics, reproductive status at the time of sterilization, difficulty of the surgery, and prior life experience explain some of these differences. The bottom line is that each animal responds individually, and must be assessed, monitored and treated as such.

Pain may be difficult to recognize in animals. Most animals are stoic and are evolved to hide signs of pain. The most consistent sign of pain in an animal is a change in behavior, or expression of unusual behaviors (Table 1). The observations of someone who knows the animal (e.g., guardian) are invaluable for assessing pain.

Signs of pain may include any of the following:

- Reduction in normal behavior, e.g., lethargy, reduced activity, depressed, not meeting members of the household at the door, not playing, failing to groom (especially in cats)
- Expression of abnormal behaviors or personality change, e.g., more aggressive, reclusive, restless, or 'clingy'. Cats usually withdraw and hide if they are feeling unwell or painful.
- Reluctance to move, or moving stiffly and awkwardly. With abdominal pain, animals may hold the abdominal muscles very tense and tuck up the abdomen and arch the back.
- Crying or trying to bite or scratch when touched
- Licking, chewing, scratching or otherwise traumatizing the site of the surgery or elsewhere on their own bodies.

Even animals who do not show overt signs of pain as described above should be evaluated daily for at least one week after surgery for evidence of pain and to assess the surgical wound.

Evaluating a patient for pain after sterilization surgery:

1) First understand the animal's normal disposition and level of anxiety. Some animals normally snap or scratch when one gets near or tries to touch them, whether they are healthy or experiencing pain. Alternatively, anxiety or stress may mask signs of pain.

2) Observe the animal as she walks, stands, sits or lies down. Does he look comfortable and relaxed? Is she anxious and nervous? Is he holding his or her body tensely, tummy tucked up, walking stiffly and gingerly? Has the cat been crouched in one position in her litter box for the last 24 hours without eating, drinking or urinating? A comfortable animal who is not in pain will lie in a relaxed position, will be alert and responsive (unless sleeping), with regular, relaxed respiratory and heart rates.

3) Before examining the animal, try to make friends with her and touch him gently on the back or scratch her under the chin or behind the ears – somewhere where you do not expect anything to hurt. If the patient is very tense and nervous and tries to snap, it may be difficult to accurately evaluate pain. Feral animals who are not accustomed to being handled may be very nervous.

4) Perform a standard physical examination that includes auscultation of heart and lungs. Elevated heart rate and respiratory rate are signs of pain, but also of stress.

5) Gently palpate the abdomen, beginning on the sides.

   a) Pressure should be minimal. A good way to know how much pressure is appropriate is by testing it on oneself. Close your eyes and push a finger against an eye. If it hurts, you are pressing too hard. Apply the same degree of comfortable pressure when examining an animal.

   b) Signs of pain include tensing of the abdominal muscles, snapping at your hand, quickly turning the head, or vocalization. If this is a strong reaction, then something is wrong and requires immediate attention. A moderate or mild response indicates the need for a day or two additional pain relief medication. A lack of response either means that the animal is too nervous to show a response, or that she is very stoic, or that he is not experiencing pain in response to your palpation. Monitor the other indicators of pain and try to reassess when the patient may be more calm.

6) Examine the area of the surgical incision. If it is swollen, inflamed or producing discharge, two things are necessary:

   a) Continue pain relief medication

   b) Figure out what is causing the inflammation and address the cause. Is it infected? Is the animal traumatizing the skin or wound? Is there a suture reaction?

All the criteria in Steps 1-6 must be considered together, in the context of the animal's normal behavior and the stress or anxiety that the animal experiences during the examination. The presence or absence of a single element does not necessarily rule out or indicate pain – unless the surgery site or the surrounding skin are inflamed. In that case, additional pain medication and addressing the underlying cause of the problem are essential.

If in doubt, it is usually better to err on the side of treating pain that may not be present, rather than risking the neglect of pain. Give pain medication and re-evaluate the animal's behavior a few hours later. If the patient is more normal, more relaxed, less reactive to being touched or handled, then he was probably in pain. If not, try to make the animal more comfortable, reduce sources of stress and anxiety, and evaluate again.

Table 1: Signs of pain*

General signs	Specific signs
Loss of normal behavior	Decreased ambulation or activity, lethargic attitude, decreased appetite, decreased grooming (cats). Harder to assess in hospital than by guardian at home.
Expression of abnormal behaviors	Inappropriate elimination, vocalization, aggression or decreased interaction with other pets or family members, altered facial expression, altered posture, restlessness, hiding (especially cats)
Reaction to touch	Increased body tension, flinching, vocalizing or snapping in response to gentle palpation of injured area and palpation of regions likely to be painful, e.g., neck, back, hips, elbows (cats) and areas recently subjected to surgery.
Physiologic parameters	Elevations in heart rate, respiratory rate, body temperature, and blood pressure; pupil dilation.

From: Hellyer, P., Rodan, I., Brunt, J., Downing, R., Hagedorn, J.E., Robertson, S.A. 2007. AAHA/AAFP pain management guidelines for dogs & cats. *Journal of the American Animal Hospital Association*, 43:235-248.

*Note that many of these are also signs of stress, particularly the physiologic parameters. See text above for interpretation of signs of pain or stress.

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Appendix 7: Vital parameters during anesthesia

	Normal (anesthetized)		Requires Treatment		Critical Level	
	Dog	Cat	Dog	Cat	Dog	Cat
Respiratory rate (breaths per minute)	8-20	10-28	< 8	< 10	< 6	< 7
Heart rate (beats per minute)	60-120	140-200	<60 or >140	<100 or >200	<40 or >175	<90 or >225
Temperature (ºC)	37.5-39.2	37.8-39.5	<37 & >39.5	<37.5 & >39.5	<36 & >40	<36 & >40
Temperature (ºF)	99.5-102.5	100-103	<99 or >103	<99.5 & >103	<97 & >104	<97 & >104
Capillary refill time (seconds)	< 2	< 2	> 2	> 2	> 3	> 3

Values may vary in animals given alpha-2 agonists: see Appendix 5: Anesthetic and analgesic drug protocols.

The respiratory rate of pediatric patients should be 2-3 times the adult respiratory rate (cf. Section 8.2: Pediatric physiology in anesthesia).

Mucous membrane color should be healthy pink. Membranes should remain moist. They should be moistened regularly during anesthesia with a damp gauze sponge. Note effects on mucous membrane color with alpha-2 agonists (Appendix 5: Anesthetic and analgesic drug protocols).

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Appendix 8: Guide for monitoring depth of anesthesia

Plane	Characteristics	Laryngeal reflexes	Respiration	Jaw & tongue	Eyes
1: Sedation	Sedation, possible disorientation	Swallowing reflex intact, can maintain own airway	Normal	Strong jaw & tongue tone	Drowsy but all reflexes intact
2: Unconscious, Delirium	• Loss of consciousness • Hyper response to stimulation • Irregular respiration • Vocalization • Uncontrolled movement • Retching, vomiting	Swallowing reflex intact, can maintain own airway	Irregular but using intercostal muscles and diaphragm	• Strong jaw tone • Tongue voluntarily retracted when pulled	Palpebral reflex intact Pupils dilated
3: Surgical	• No vocalization or voluntary movement • Muscle relaxation • Analgesia, but painful stimuli cause ↑HR, RR, BP	See sub-planes	See sub-planes	• Loss of jaw tone • Tongue relaxed, does not retract when pulled	Pupillary constriction
Surgical sub-plane 1	• Light surgical anesthesia • Sensitive to pain	Swallowing reflex intact, can maintain own airway	Regular with strong chest movement & diaphragm function	• Minimal or no jaw tone • Tongue relaxed, does not retract when pulled	• Palpebral reflex intact • Eyeball centrally positioned
Surgical sub-plane 2	Best plane for most surgeries, except for particularly painful surgeries	Loss of swallowing reflex, must protect airway	Regular with strong chest movement & diaphragm function	• No jaw tone • Tongue relaxed	• Loss of palpebral reflex • Pupils fixed (usually central) • Eyeball usually rotates down
Surgical sub-plane 3	• Deep surgical anesthesia: for very painful surgeries • Decreased BP	No swallowing reflex, must protect airway	• Diaphragmatic breathing only • Breathing shallow • Assist ventilation	• No jaw tone • Tongue relaxed	• No palpebral reflex • Pupils fixed • Eyeball usually central • Cornea dry (no lacrimation)
Surgical sub-plane 4	Too deep, crisis imminent	No swallowing reflex, must protect airway	• Requires assisted ventilation • Respiration close to ceasing altogether	• No jaw tone • Tongue relaxed	• No palpebral reflex • Pupils fixed • Cornea dry
4: Medullary depression	• Anesthetic crisis: death imminent • Pulse weak (BP too low) • Reverse anesthesia and begin cardio-pulmonary support immediately.	No swallowing reflex, should be intubated	Respiratory arrest	• No jaw tone • Tongue relaxed	• No palpebral reflex • Pupils dilated • Cornea dry

Note that in Plane 2 of anesthesia (during induction and arousal), animals may be hyper-responsive to stimuli, but may not have good motor control. During this plane of anesthesia, it is critical to avoid stimulation of the animal (sounds, movements, light) and to protect the animal from injuring him or herself. Refer to Appendix 5: Anesthetic and analgesic drug protocols and Appendix 8: Guide for monitoring depth of anesthesia.

Appendix 9: Emergency drugs quick reference drug chart

Drug	Use	Dose	Route
0.9% NaCl, Lactated Ringer's, or Hartmann's	Fluid therapy – shock, rapid rehydration	60-90 ml/hr (dog) 40-60 ml/hr (cat)	IV
Adrenaline 1:1000 = 1 mg/ml Adrenergic agonist	cardiopulmonary arrest	0.01 mg/kg 0.1-0.2 mg/kg	IM, IV, IT IT dose double IV; dilute in 1-3 ml saline
	Anaphylactic shock	0.01 mg/kg	IM, IV
Atropine	Bradycardia (take care with alpha-2 agonists)	0.02-0.04 mg/kg	SQ, IM, IV
Calcium gluconate, 10%	Hypocalcemia, e.g., eclampsia	0.5-1.5 ml/kg	IV slowly, to effect. Monitor heart for arrhythmia & bradycardia.
Dexamethasone	Anaphylaxis Prednisolone is preferable	0.25-1.0 mg/kg	IV
Diazepam or midazolam	Seizures	0.25 - 0.5 mg/kg (IV) 2 mg/kg rectal	IV or rectal, to effect
Diphenydramine Antihistamine	Anaphylaxis	2.0 mg/kg	Oral, SQ, IM
Doxapram	Respiratory stimulant. Combine with manual ventilation for respiratory arrest.	Adult & pediatric: 5 mg/kg. Repeat in 15-20 minutes as needed	IV slowly, or IM
		Neonate: 1-5 mg/puppy 1-2 mg/kitten	SQ, into umbilical vein or sublingual
Glucose or Dextrose, 5%	Hypoglycemia	20 ml/kg	IV slowly

Drug	Use	Dose	Route
Hydrogen peroxide, 3%	Induction of vomiting*	~0.5 ml/kg	orally
Lidocaine	Ventricular arrhythmia (verify with ECG)	dog: 1-2 mg/kg cat: 0.2-1 mg/kg	IV or 2x dose IT
Prednisolone (injectable)	Anaphylaxis Preferably use this instead of dexamethasone.	2-4 mg/kg	IV
Sodium bicarbonate 8.4% solution = 1 mmol/ml = 1 mEq/ml	Metabolic acidosis associated with cardiac arrest	1.0 mEq over 1-2 min, then 0.5 mEq at 10 min intervals as needed	IV slowly
Terbutaline	Bronchodilator, e.g., for anaphylaxis or feline asthma	0.01 mg/kg	SQ, IM,IV

*H_2O_2: If possible, use xylazine to induce vomiting in cats, and apomorphine eye drops for dogs instead of H_2O_2.

Refer to Appendix 11: Emergency treatment of cardiac and respiratory arrest for cardiopulmonary crisis protocols

Appendix 10: Emergency treatment kits

An emergency treatment kit, together with dosage charts, should be stored in a secure location and must be readily available within the surgical facility. Refer to Appendix 9: Emergency drugs quick reference drug chart.

The kit should be checked and re-stocked after each use. The date of the checks should be recorded on the kit. The responsibility for doing this should be assigned to a specific individual (in most instances a veterinarian or a veterinary nurse).

The emergency kit should be used for immediate intervention treatment of shock, cardiac distress, respiratory distress, seizures, anaphylactic reactions or other emergency situations.

Drug and supply stock for Emergency Kit

Drugs

Adrenaline
Antihistamine (injectable)
Alcohol (75% isopropanol)
Anesthetic drugs
Atropine
Butorphanol or buprenorphine
Calcium gluconate 10%
Charcoal (activated, for poisoning)
Diazepam
Diphenhydramine
Doxapram
Euthanasia solution
Flea spray
Fluids (Saline (0.9% NaCl), LRS or Hartmann's)
Glucose 50% or 5%
Heparin (for flushing IV catheter)
Hydrogen peroxide 3%
Iodine / Betadine
Lidocaine
Prednisolone or Dexamethasone, injectable (for analphylactic reactions)
Reversal drug for anesthetic (e.g., atipamezole, yohimbine, naloxone)
Sodium bicarbonate
Terbutaline (injectable)
Wound ointment (silver sulfadiazine or triple antibiotic)

Veterinary supplies

Chorhexidine, 4%
Cotton balls
Cotton buds (Q-tips)
Cotton roll
Dog treats (e.g., dried liver strips)
Endotracheal tubes, assorted sizes
Eye lubricant (ointment)
Fluids giving sets (for IV fluids)
Food, dry: dog & cat
Gauze squares
Gauze bandage
Gloves, latex examination, S, M, L
IV catheters: 24, 22, 20G
Injection port caps for IV catheters
Lubricant for endotracheal tubes
Needles: 22, 20, 18G
Rabies vaccine
Surgery pack (basic laceration pack, autoclaved)
Suture material, absorbable, 0, 2-0, 3-0
Syringes: 1, 3, 5, 10, 20, 60 ml
Tape (bandage): 1 cm & 2.5 cm wide
Water (drinking quality), 2L
VetWrap® or similar bandaging material

Equipment

Ambu bag
Batteries for flashlights & laryngoscope
Bowls for drinking water
Calculator
Cat cages
Y-pole, catch pole
Clippers, charged
Cloth sack, e.g., pillow case (for catching/holding cats)
Dog cages
Drug dosage charts for quick reference
Duct tape
Flashlights or head lamps
Glucometer
Hemostat forceps, for general use
Laryngoscope
Leashes and collars
Muzzles or rope to fashion muzzles
Net for catching dogs / cats
Pens and permanent-marker pens
Plastic rubbish bags
Record forms & clipboard
Refractometer
Rubbish bin (closable box)
Scissors (multipurpose)
Sharps container (empty water bottle with cap)
Stethoscope
Thermometer (rectal)
Towels or fleeces
Tube for gastric lavage

Notes for drug list

Refer also to:

Appendix 9: Emergency drugs quick reference drug chart

Appendix 11: Emergency treatment of cardiac and respiratory arrest

Appendix 12: Emergency treatment for anaphylactic reaction

1. Adrenaline (adrenergic agonist) 1:1000 solution (1 mg/ml).

   Cardiac arrest and anaphylactic shock normally require a dose of 0.1 - 0.2 mg/kg, given IV or intratracheally.

2. Atropine

   For the treatment of bradycardia, but with care when used with alpha-2 adrenergic drugs (see notes on specific drugs in Appendix 5: Anesthetic and analgesic drug protocols).

3. Calcium gluconate

   For treatment of hypocalcemia, e.g., eclampsia.

4. Diazepam or midazolam

   For control of seizures: 0.25 - 0.5 mg/kg as intravenous bolus. Wait for 5 minutes, if seizures persist, repeat the bolus.

   Diazepam can be given rectally if intravenous access is not possible. Avoid giving diazepam IM. Use midazolam instead.

5. Diphenhydramine (antihistamine)

   Following initial emergency injectable dose, subsequent oral doses may be given at 2 mg/kg PO as often as every 8 hours if needed. (Human tablets are usually 25 mg.)

   Cats may demonstrate paradoxical excitement in response to diphenhydramine.

6. Doxapram (respiratory stimulant)

   For treatment of respiratory suppression or respiratory arrest during anesthesia. Combine with positive-pressure ventilation with an anesthesia machine or with an ambu bag.

   Administer IV slowly (over 3-5 minutes) or IM, 5 mg/kg

7. Fluids: 0.9% NaCl (isotonic saline), Lactated Ringer's solution or Hartmann's solution

   Fluids should be administered as a continuous intravenous infusion at controlled rates, via a securely placed sterile intravenous catheter.

8. Glucose or dextrose

   Dextrose (glucose) solution 5%: best added to crystalloid fluids (0.9% NaCl, Lactated Ringer's solution or Hartmann's solution).

   Glucose is given to treat, or to prevent, hypoglycemia. It is not used for treatment of hypovolemic or toxic shock.

   May deliver up to 40-50 ml/kg every 24 hours intravenously; monitor requirement with blood glucose measurements.

9. Hydrogen peroxide (H_2O_2)

   For induction of vomiting in case the patient has ingested something poisonous. Xylazine (0.5 mg/kg IM or IV) may be used instead (works particularly well in cats), or apomorphine eye drops for dogs.

10. Lidocaine

    For treatment of ventricular arrhythmias. Diagnosis should be accurate, ideally verified by ECG.

11. Prednisone or prednisolone, injectable.

    For treatment of anaphylactic reactions, e.g., vaccine reaction or penicillin reaction.

    Short-acting glucocorticoids such as prednisone or prednisolone are preferred for treatment of anaphylaxis. If these are not available, use dexamethasone instead (long-acting).

    Note that glucocorticoids are no longer recommended for treatment of shock. Solu-Delta Cortef® (prednisolone sodium succinate) may be recommended for treatment of spinal trauma, if treatment is initiated within 12 hours of the traumatic event (1-2 mg/kg IV). Glucocorticoids are contraindicated in head trauma. Solu-Delta Cortef® is purchased as a lyophilized powder that can be stored only for 3 days after it is reconstituted, or stored in frozen aliquots at or below -20 C immediately following reconstitution.

12. Sodium bicarbonate 8.4% solution (8.4% = 1 mEq/ml = 1 mmol/ml))

    For treatment of metabolic acidosis associated with cardiac arrest or other crises.

    Do not use with Lactated Ringer's solution.

    The administration of an alkalinizing agent may not be efficacious for short-duration cardiac arrests but may improve survival in long-duration cardiac arrests.

    Recommendation in the treatment of cardiac arrest: give no sodium bicarbonate for the first 5 to 10 minutes. Then administer 0.5 mEq/kg per 5 minutes' duration of cardiac arrest, or add to IV fluids at 1-2 mEq/kg. If it is known or suspected that metabolic acidosis predated the cardiac arrest, then the bicarbonate dosing should start right away.

13. Terbutaline (beta-adrenergic agonist, bronchodilator)

    For use in allergic reactions and for feline allergic bronchitis (feline bronchial asthma syndrome requires emergency intervention).

    Once the crisis has been managed and the animal is stable, terbutaline can subsequently be given orally if needed (e.g., asthmatic cats).

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Appendix 11: Emergency treatment of cardiac and respiratory arrest

Heart beat slow (ca. ½ normal rate)

1. Administer atropine 0.04 mg/kg IV or IM
2. Monitor heart rate, rhythm and pulse carefully
3. Monitor respiration carefully and treat as needed.
4. Keep patient warm

Heart beat very slow (less than ½ normal)

1. Administer adrenaline (1 mg/ml)
   a. IV route: 0.01 ml/kg diluted in 5 ml saline
   b. via ET tube: Insert clean rubber catheter or IV tubing to level of carina.
      - Inject 0.02 ml/kg adrenaline (double IV dose), followed by 1-3 ml saline
      - Give 2 strong ventilations with ambu bag or oxygen from machine
2. Commence IV fluids at 2 ml/kg/hr
3. Monitor heart beat and pulse carefully
4. Monitor respiration carefully and treat as needed.
5. Keep patient warm

Breathing slow or shallow

1. Intubate
2. If on gas anesthesia, turn off anesthetic.
3. Start artificial respiration with ambu bag or anesthesia machine: 10 breaths /minute. Neonates: 20-40 breaths per minute, gently.
4. Provide supplemental oxygen if possible.
5. Monitor heart rate, pulse and respiration carefully
6. Keep patient warm.

Breathing stopped

1. Intubate
2. If on gas anesthesia, turn off anesthetic.
3. Start artificial respiration with ambu bag or anesthesia machine: 10-20 breaths /minute. Neonates: 20-40 breaths per minute, gently.
4. Provide supplemental oxygen if possible.
5. If no response within two minutes:
   a. Administer doxapram IV (5 mg/kg, administer slowly over 3-5 minutes) while continuing artificial respiration.
   b. If tranquilized with alpha-2 adrenergic drugs (xylaxine, medetomidine, dexmedetomidine) or with an opiate, administer the appropriate reversal drug. Be prepared for immediate arousal.
   c. Continue manual ventilation at 10-20 breaths per minute
6. Commence IV fluids at 5 ml/kg/hr
7. Continually monitor heart rate and strength of pulse
8. Keep patient warm

Heart beat and breathing have stopped. This is CARDIOPULMONARY ARREST.

1. Remember ABC:

   AIRWAY: animal should already be intubated.

   BREATHING: ventilate manually or with machine.

   a. 10 breaths per minute

   b. Pressure: 10-15 cm H_2O

   c. Tidal volume: 10 ml/kg

   CARDIAC: cardiac compressions.

   a. Place patient in lateral recumbency (right or left)

   b. Compress chest 80-100 times per minute (dog); 100-150 (cat)

   c. Compress rib cage 1/3 to 1/2 way, and allow it to expand fully before next compression.

   d. Coordinate ventilation with compressions: inhalation during chest expansion phase.

   e. If one person is doing both compression and ventilation: 30 compressions, 2 breaths, 30 compressions, 2 breaths, etc.

2. Administer adrenaline (concentration: 1 mg/ml)

   a. IV 0.1 ml/kg.

   -- Follow IV adrenaline with a bolus of fluids (1-5 ml).

   -- If CPR is continued 10 minutes without response, repeat adrenaline bolus.

   b. Intra-tracheal: Insert clean rubber catheter or IV tubing through the ET tube to the level of the carina.

   -- Inject 0.2 ml/kg adrenaline, followed by 1-5 ml sterile saline

   -- Give 2 strong ventilations with ambu bag or oxygen from machine

   c. Intra-cardiac injection if there is no pulse at all, at 0.1 ml/kg

   d. Repeat every 4 minutes until heart is beating again

3. IV fluids

   a. Dogs 20 ml/kg/hour

   b. Cats: 10 ml/kg/hour

4. Sodium bicarbonate

   a. Metabolic acidosis often begins ca. 10 minutes after resuscitation, particularly if resuscitation has taken a long time.

   b. Best to monitor blood pH and electrolytes, but if that is not an option, administer $NaHCO_3$ at 1-2 mEq/kg into IV fluids

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Appendix 12: Emergency treatment for anaphylactic reaction

Acute anaphylaxis may follow vaccination, administration of certain drugs (e.g., penicillin), insect stings or other allergens. Reactions may occur within minutes or over the following few days. When vaccinating animals, one should monitor them for 15 minutes to make sure that they will not have an acute anaphylactic response. Clinical signs include:

- Dyspnea
- Vomiting
- Diarrhea
- Abdominal pain
- Hypersalivation
- Cyanosis (blue mucous membranes)
- Swollen face, tongue or throat
- Shock (pale mucous membranes, capillary refill time >2 sec, bradycardia (particularly in cats) or rapid, thready pulse, weak peripheral pulse, altered consciousness or loss of consciousness)
- Death

Treatment

1. Adrenaline at 0.01 – 0.02 mg/kg IV or IT (intra-tracheal) if the animal is having trouble breathing, or if the face or throat are swelling rapidly.
2. Response to adrenaline should be seen immediately. Repeat adrenaline dose if the animal is still showing signs of distress 5 minutes after first injection. If the adrenaline was given IM, the expected response will be slower.
3. Adrenaline may result in cardiac arrhythmias, particularly following multiple or high doses.
4. Prednisolone sodium succinate (Solu-Delta Cortef® 2-4 mg/kg IV, one dose) or dexamethasone (0.25-1 mg/kg IM, one dose).
5. Diphenhydramine (antihistamine): 1-2 mg/kg IM
6. If reaction is severe or if dog is showing evidence of cardiovascular collapse (shock), administer IV fluids (NaCl or LRS). Cat: 40-60 ml/kg. Dog: 60-90 ml/kg.
7. If the animal is severely dyspnaeic: aminophylline (4-8 mg/kg IM or slow IV) or terbutaline (0.2 mg/kg IV or IM)
8. Prevent the development of gastric ulcers that often occur after a massive release of histamine, particularly in dogs.
   - Follow treatment for anaphylaxis with a few days of sucralfate (0.5-1 gram, every 6-12 hrs) or famotidine: 0.5 mg/kg PO, IM, SQ, every 12-24 hours.
   - If vomiting intermittently, use ranitidine (0.5 mg/kg IM, BID) or metoclopramide (0.2 mg/kg SQ, BID).

Prophylaxis

1. Animals with a history of anaphylactic reactions to vaccines may be pre-treated 20-30 minutes prior to administration of the vaccine.
2. Diphenhydramine: 1-2 mg/kg SQ or IM.
3. Dexamethasone: 0.25 mg/kg SQ or IM
4. Monitor the animal closely for 15 minutes after vaccination and be prepared to intervene with emergency treatment as outlined above.

Appendix 13: Supply list for field sterilization events

The following is a recommended list of supplies needed for field spay/neuter projects, including drugs, consumable veterinary supplies, and equipment. The list takes into consideration potentially challenging conditions, e.g., where running water may not be available, and in which case surgical drapes and instruments may need to be washed on site for re-sterilization. Electricity must be available if surgical instrument packs are to be washed, repacked and autoclaved on site. Specific items may be omitted or added to this list for various areas or situations.

Drugs

Adrenaline

Antihistamine (injectable)

Anesthetic drugs

Antibiotic, broad-spectrum, long-acting, e.g., 3- or 5-day penicillin

Atropine

Calcium gluconate 10%

Charcoal, activated, for incidental poisoning cases that may be presented on site

Diazepam

Diphenhydramine

Doxapram

Drugs to treat commonly-seen conditions other than common parasites and minor wounds

Ear cleaning solution

Ear ointment – bacterial / fungal infection

Euthanasia solution

Eye lubricant (artificial tears ointment)

Flea spray

Fluids (Saline (0.9% NaCl), LRS or Hartmann's)

Glucose 50% or 5%

Heparin (for flushing IV catheter)

Hydrogen peroxide 3%

Lidocaine – emergency drug & cat intubation

Metoclopramide or ranitidine

NSAID or other long-acting analgesic for post-operative pain management, injectable

Parasite treatment drugs: endoparasites and ectoparasites. Refer to Appendix 4: Anti-parasitic drugs

Povidone iodine / Betadine

Prednisolone or Dexamethasone, injectable (for penicillin or vaccine reactions)

Pre-medication drugs (cf. Appendix 5: Anesthetic and analgesic drug protocols)

Reversal drugs for anesthetic (e.g., atipamezole, yohimbine, naloxone)

Skin glue

Sodium bicarbonate

Terbutaline (injectable)

Vaccine – rabies, feline multiple, canine multiple

Wound ointment (silver sulfadiazine or triple antibiotic)

Veterinary supplies

Alcohol (75% isopropanol)

Ambu bags

Autoclave indicator tape

Bandaging material, e.g., VetWrap®

Benzalkonium 5%, for cold sterilization & hand scrub, or chlorhexidine 4%

Caps for surgery (cloth or disposable)

Chlorhexidine 4% for cold sterilization and pre-surgical hand scrubbing, or benzalkonium

Bleach (sodium hypochlorite) for washing blood stained cloths & sterilization of inanimate surfaces

Cotton buds (Q-tips)

Cotton rolls or balls

Endotracheal tubes, sizes 3.0 - 12

Gauze bandage rolls

Gauze squares – sterile and non-sterile

Gloves, non-sterile examination gloves, sizes small, medium, large

Gloves, sterile surgical gloves, appropriate sizes

Gown, surgical (sterile cloth or disposable)

Injection port caps for IV catheters

IV catheters, 24G, 22G, 20G

IV fluid giving sets. If working with cats and small dogs, these must include a container that allows filling of ~200 ml (pediatric giving sets).

Laryngoscope with 2 or 3 different blade sizes

Lubricant for endotracheal tubes. (Sterile lubricant sold for use with condoms and other „personal use". Must be free of perfumes or dyes.)

Nail clippers

Needles, 18G, 22G, 24G

Scalpel blades (#10 or 15)

Space heater for surgery and recovery areas in cold temperatures

Stethoscopes

Styptic powder

Surgery packs (cf. Appendix 2: Basic instrument pack for spay and neuter surgery)

Surgical drapes, sterile cloth or disposable

Suture material, 3-0, 2-0 and 0 size range usually suffices for dogs and cats

Syringes (1, 3, 5, 10, 20, 60 ml)

Tape, bandaging 1 cm & 2.5 cm

Thermometers, rectal

Trays, stainless steel, for cold sterilization & instrument washing

Water: drinking water for dilution of disinfectant, washing surgeons' hands if tap water is not of drinking quality

Water for washing instruments, drapes, tables, etc. A running water source is ideal, but may not always be available.

Equipment & general supplies

Autoclave machine

Basin for washing surgical drapes, towels, hands

Batteries, various sizes for relevant equipment

Pots for alcohol and iodine swabs

Blankets or large towels for bedding in cages. In dire situations, can at least use cardboard to place under animals.

Bowls for animals to eat & drink

Calculators

Camera

Y-pole, catch pole

Cloths for cleaning tables and other surfaces

Clip boards

Clippers with chargers

Clipper blades, spare, clean

Clock with second hand for surgery & examination room areas

Cloth sack (e.g., pillow case or similar), for restraining cats

Clothes line and clothes pins for drying surgical laundry

Clothing for veterinary staff – scrub tops, scrub trousers

Cooler or refrigerator for vaccine

Cages: various sizes as needed

Duct tape

Fan for animal holding area, in hot temperatures

Flash light: small & bright, for improving surgeon's visibility & intubation

Food for dogs & cats

Food & drink for staff

Freezer packs to keep vaccine cool

Gas anesthesia machine

Gasoline, if reserve may be needed for vehicles

Heating pads, warm water bottles, microwavable sacks of rice or lentils or other individual heat source for each surgery table + additional for recovery area

IV fluid stands

Hemostat forceps for general use

Head lamps for surgical staff

Lamp, surgical; one per surgery table

Leashes & collars, assorted sizes

Leather gauntlets or gloves for handling fractious cats

Measuring tape

Microwave oven, if using microwavable heat source bags or to warm fluids in cold temperatures

Mobile phones or 2-way radios

Muzzles, or rope to fashion muzzles

Net for catching dogs & cats

Oxygen tank (1 per anesthesia machine, or at least 1 for emergency use)

Pens: writing pens & marking pens (permanent marker)

Pipe cleaners, for washing endotracheal tubes

Pitchers for diluting disinfectant, with lid

Pulse oximeter, capnograph and other monitoring machines, if available

Record forms (cf. Appendix 14: Examples of clinical forms and record sheets)

Rope or cloth strips of very soft material, to tie limbs for positioning of patient on surgery table

Rubbish baskets (one per surgery table + additional)

Rubbish bin liners (plastic bags – small and large)

Scissors, general purpose

Scrub brushes or sponges for surgeon hand scrubbing

Sharps containers (empty plastic water bottles will do; keep caps!)

Surgery tables

Sink with running water (or basins if no running water)

Spray bottles for disinfectant

Towels or fleeces, to place under patients on surgery tables. In dire situations, can at least use cardboard to place under animals.

Tray for holding each patient's medication - 14x14 cm square plastic dish works well

Treats for dogs & cats

Vacuum: small hand-held vacuum cleaner for clipped fur

V-tray or cushions with washable surface, for maintaining animal's position during surgery. A warm water bottle or warmed bean bag on either side of the patient works well for this.

Washing powder, if surgical laundry is washed on site

Water drums if water is carried to location

Weigh scale: must be able to read up to 50kg, in 0.1kg increments

Appendix 14: Examples of clinical forms and record sheets

1. Guidelines for caring for your pet before and after spay/neuter surgery
2. Clinical Evaluation Record (physical examination form)
3. Anesthesia record

Guidelines for caring for your pet before and after spay/neuter surgery

Preparing your dog or cat for spay/neuter surgery:

- The veterinarian will check your pet before surgery to make sure he or she is healthy, but your observations about your pet are very important. Be sure to tell the veterinarian of any changes in behavior, appetite, activity, urination, defection or anything else that you think may be abnormal.
- If your pet has never had vaccinations, s/he should be vaccinated two weeks before the surgery. If this is not possible, s/he should be vaccinated on the day of surgery.
- If possible, bathe your dog or cat on the day before surgery, and keep him or her clean and warm.
- If your dog or cat is pregnant or in estrus, she may be spayed if she is healthy. The veterinarian should discuss this with you.
- On the day of surgery:
    - Dogs and cats 6 months or older: do not give any food for eight hours before the surgery.
    - Puppies and kittens less than 4 months old: allow to eat and drink normally.
    - Make sure that the animal always has access to clean drinking water. Do not withhold water before surgery.

Taking care of your pet after spay/neuter surgery

- Provide a warm, soft, quiet area for your pet to rest after surgery. Your pet might seem a bit quiet for a day or two, and that is normal. Give him or her time to rest and sleep, and lots of love.
- Feeding dogs and cats 6 months or older: provide water as soon as your pet has recovered from anesthesia. Offer half of a normal meal 4 hours after s/he wakes up. Offer normal amounts of food beginning the day after surgery. Do not force your pet to eat or drink. Simply provide the choice for him or her to do so.
- Feeding dogs and cats less than 4 months old: offer food and water as soon as he or she has woken from anesthesia and is able to stand up by him or herself. The food should be soft and easy to eat.
- Provide drinking water as soon as the animal has woken from anesthesia and is able to stand by him or herself. Drinking water must always be available to your pet.
- Sutures will dissolve on their own in a few weeks. They do not have to be cut out.
- Do not touch the incision site or wash it unless instructed to do so by the veterinarian.
- If the veterinarian sent medicine home with you, give the medicine every day according to instructions.
- Monitor the animal daily for weakness, lethargy, vomiting, diarrhea, constipation, difficulty voiding urine, pain, bleeding, and any abnormal behavior. Call the veterinarian immediately if you see any of these changes.
- Check the surgical area daily for signs of swelling, redness, discharge, the incision opening or sutures coming out. Call the veterinarian immediately if you see any of these changes.
- Prevent your pet from licking or scratching the surgical area. The veterinarian can provide Elizabethan collars or medicine if necessary. Call the veterinarian immediately if the animal is scratching or licking a lot, if the skin is pink or swollen or wet, or if the sutures are coming out.
- Prevent your pet from being too active and jumping for 7 days following surgery. Walk your dog only on a leash (do not allow him or her to run loose). Keep your cat indoors.
- Do not bathe your pet or allow him or her to go swimming for 2 weeks following surgery.

Clinical Evaluation Record

Date :____________________ Species:____________________ Breed: ____________________ Sex: __________

Weight: ___________kg ❏ estimated ❏ actual Birth date: ____________________ ❏ estimated ❏ actual

Description:___________________________________ Animal ID/name:__________________________________

Veterinarian:__________________________________ Guardian: ______________________________________

History (where & when found, previous illness, any other known information)

Physical examination Temp _____ C

Attitude (e.g., alert, depressed, aggressive, lethargic, unresponsive, unconscious, fearful, anxious)

General appearance (e.g., emaciated, thin, fat, good body condition, healthy, sick, mangy, soiled)

Skin (e.g., healthy, inflamed, raw, describe lesions)

Fur (e.g., thick, sparse, dull, shiny, patchy, clumps falling out)

Eyes (e.g., discharge (describe), inflamed, sunken, protruding, lesion on cornea, holding eye closed or blinking too much, eyeball twitching, wounds, symmetry, eyelashes rubbing against cornea, eyelid rolled inward or drooping, pupillary light reflex)

Left Right

Ears (e.g., smell, dirty, wax, discharge (describe), wounds, swelling in ear canal or pinna)

Left Right

Mouth & gums (e.g., color, dry/moist, smell, inflamed, wounds, capillary refill time)

Teeth (e.g., fractured, missing, exposed dental canal, tartar)

Right upper
Right lower
Left upper
Left lower

Lymph nodes (size & firmness: submandibular, pre-scapular, axillary, sub-lumbar, inguinal, popliteal)

Digestive system (e.g., appetite, color & quality of faeces, vomit, bloated abdomen, flatulence, burping, abdominal masses)

Respiratory system (e.g., cough, nasal discharge (describe), breath sounds, difficulty breathing) RR _____/min

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Cardiovascular system (heart rate, rhythm, murmurs, pulse strength, pulse coordination with heart beat) HR_____bpm

Legs and joints (e.g., fractures, joint mobility, paw pads, wounds, nails)

Nervous system (e.g., convulsions (describe detail, frequency, duration), tremors, twitching (which body part?), head tilt, poor balance, pressing head against wall, weak (which body part?), paralyzed (which body part?)

Urinary & genital systems (e.g., wounds or discharge from penis or vulva, quality and frequency of urine, presence of both testicles, growths in vagina or on penis, female reproductive status)

Scars, wounds, lumps (e.g., location, size, what they look like)

Assess hydration status

Dehydration	Mucous membranes	Loss of skin turgor	Eyes	Pulse	Consciousness
4-5%	Slightly dry	Mild	Moist, normal	Strong	Normal
6-7%	Dry	Moderate	Moist, normal	Strong	Normal
8-10%	Dry	High	Dry, retracted	Weak, rapid	Weak, depressed
12% +	Very dry	Complete	Severely retracted	weak, rapid	Unconscious or abnormal

Diagnosis of illnesses (list possibilities)

Laboratory tests (type of test, test result, test date, where test was done)

Medications & treatment given (use fluid calculation sheet to calculate fluid requirement):

Drug name	Concentration	Amount given	Route	Time

Additional medical & nursing care required: (type, frequency, duration)

Companion Animal Anesthesia Form

Animal name & ID____________________ Sex ❏ M ❏ F Date ____________

Species ____________ Breed__________ Body wt._______kg ❏ actual ❏ estimated

Description ____________________ Birth date ____________

Guardian____________________ Tel:____________

Surgeon________________ Assistants________________

Attitude: ❏ calm ❏ excited ❏ aggressive ❏ anxious

History & reason for anesthesia: ____________________________

____________________________ Fasting time ______ hours

Anesthetic drugs

Drug name	mg/ml	Amount given (ml)	Route (IM, SQ, IV)	Time given	Effect*

Other drugs

Drug name	Dose: mg/kg	Concentration: mg/ml	Amount given (ml)	Route (IM, SQ, IV)	Time given

Fluids: Type____________ start time________ end time ________ total volume_____ ml

Intubation time __________ ET tube size _______ Tube out time __________

Recovery

Head up time __________ Time sternal _______ Time standing __________

Smooth ❏ Slow ❏ rapid ❏ Difficult (describe) ____________________

Emergency drugs

Adrenaline (1 mg/ml, 0.1 mg/kg)_____ ml IV, IT

Atropine (0.5 mg/ml, 0.04mg/kg)_____ ml IM, IV

Doxapram (20 mg/ml; 5mg/kg) _____ ml IM, slow IV

Diazepam (5 mg/ml; 0.5 mg/kg) _____ ml IV

* Effect: 0 = no effect 1= sedation 2= heavy sedation 3= surgical anesthesia 4= deep anesthesia 5= death

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Animal name ______________________________ ID ________________ Date ____________________

Isoflurane																									
Time:	:00	:10	:20	:30	:40	:50	:00	:10	:20	:30	:40	:50	:00	:10	:20	:30	:40	:50	:00	:10	:20	:30	:40	:50	:00
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10																									
5																									
0																									

Notes

Surgery start____________________

Surgery end____________________

Key

HR △

RR O

sp02 *

Appendix 15: Disinfectants

Important principles

When cleaning a room or cage with disinfectant, always be sure to remove the animal before you clean. Return the animal to the cage or room once all five steps have been completed.

Disinfectants should be allowed contact time of at least 10 minutes before being rinsed with water. Some disinfectants require longer contact times. Be sure to read instructions on the container.

Following disinfection of equipment, surfaces must be rinsed well with water before an animal comes into contact with them.

Avoid use of phenolic products (e.g., Dettol®) for cat areas, as these products are toxic to cats.

Disinfection of cages, floors, tables, or other surfaces is done in five steps.

1. Remove the animal from the cage or room.
2. Remove all organic material (faeces, urine, blood, vomitus, soil, food, etc.) and use water to rinse away anything that is still stuck to the surface. This is essential, as the disinfectant will not work properly in the presence of organic material.
3. Apply the disinfectant. Cover the entire surface, making sure to get into cracks and corners. Leave the disinfectant on for the required contact time for the disinfectant to work. For most chemicals, this is 10 minutes.
4. Rinse away the disinfectant with clean water.
5. Dry the surface – air dry or with a cloth or paper.

Properties and uses of common disinfectants

Benzalkonium chloride, 2.7-3.3% stock concentration.

- Quaternary ammonium compound
- For disinfection of equipment, e.g., cages, floors, endotracheal tubes, feeding tubes, chemical sterilization of surgical instruments, use at 1:500 = 33 parts stock solution + 467 ml water (total 500 ml)
- For use as skin scrub, use at 1:750 = 22 parts stock solution + 478 ml water (total 500 ml)

Chlorhexidine gluconate

- For hand scrub before surgery, use the 4% soap preparation.
- For cleaning countertops, floors, walls, surgery tables, etc., use as a 2% dilution, diluted in clean tap water.
- For cleaning an animal's skin prior to surgery, use 2-4% solution (not the soap preparation). Dilute in water that is clean enough to drink.
- For cleaning wounds, use 0.5% solution, diluted in sterile water or drinking water. Be sure to rinse with sterile water after using chlorhexidine – it should not be left on skin or mucosa.
- Chemical sterilization of instruments and endotracheal tubes:
  1. Wash instruments or tubes thoroughly in cold water to remove all organic material. A toothbrush works well for instruments; pipe cleaners or laboratory brushes are needed to clean the inside of endotracheal tubes.
  2. Prepare a mixture of 3 parts chlorhexidine (4%, not containing soap) and 1 part alcohol (75% isopropanol). For example: 75 ml chlorhexidine + 25 ml alcohol.
  3. Soak instruments or tubes for at least 15 minutes.
  4. Rinse thoroughly with water before use. Endotracheal tubes may be rinsed in drinking water. Instruments must be rinsed with sterile water or sterile saline (0.9% NaCl) if they are to be used for sterile procedures.

Chlorine bleach (sodium hypochlorite, 8-11% w/v NaOCl = 8-11g/L available chlorine).

- Use only on inanimate surfaces: floors, counter tops, tables, walls, cages surfaces.
- Do not use for instruments, endotracheal tubes, feeding tubes or other items that contact an animal's mucosa or internal structures. Never apply bleach to an animal's body directly.
- Dilutions:
  - Quarantine, Isolation and Surgery rooms: 50 ml/L water (1:20)
  - To decontaminate ringworm areas: 100 ml/L water (1:10)
  - Food utensils: 30 ml/L water (1:33)
  - Laundry: 10 ml/L water (1:100)
  - General areas, floors: 30 ml/L water (1:33)
- Bleach is quickly inactivated when it comes in contact with organic material. Soil, mud, feces, etc., will inactivate bleach.
- Bleach in foot baths
  - Change foot bath as often as necessary to keep it clean from organic material.
  - Place another foot bath containing only water in front of the bleach footbath. Here, soil can be removed before the shoes / boots are placed in the bleach foot bath.
  - Bleach will corrode leather, cloth and rubber. Use only for plastic boots.

Deciquan, stock concentration 10%.

- Glutaraldehyde compound
- Use only on inanimate surfaces: floors, counter tops, walls, tables, cage surfaces.
- Do not use for instruments, endotracheal tubes, feeding tubes or other items that contact an animal's mucosa or internal structures. Do not apply to the animal directly.
- Use at final dilution of 0.15 – 0.5%: dilute stock solution at 7.5 – 25 ml in 500 ml water.

Dettol® (chloroxylenol 4.8%)

- Phenolic compound
- Dilute at 1:20 (50 ml stock solution in 1 liter water).
- Appropriate for Quarantine or Isolation Units
- Use only on inanimate surfaces: floors, counter tops, tables, walls, cages surfaces.
- Do not use for instruments, endotracheal tubes, feeding tubes or other items that contact an animal's mucosa or internal structures. Do not apply to the animal directly.
- Do not use in areas housing cats.

F10

- Quaternary ammonium compound. Effective against bacteria, fungi, some viruses and fungal spores.
- Used for cleaning inanimate surfaces.
- May be used medically in nebulizers (e.g., to treat respiratory aspergillosis) at recommended dilutions.
- Use according to manufacturer's instructions.
  1. For general disinfection: 1:500 dilution (2 ml per liter of water).
  2. Disinfection against bacteria, fungi and some viruses: 1:250 or 1:125 dilution (4-8 ml per liter of water)

Gluteraldehyde

- For cold sterilization of endotracheal tubes or surgical instruments.
- Use per manufacturer's instructions.

Povidone iodine (e.g., Betadine®)

- 7.5% w/v solution
- May be used as hand scrub if surgeon is allergic to chlorhexidine, or if 4% chlorhexidine is not available.
- May be used as a skin scrub on the surgical area prior to surgery. Usually alternated with alcohol (see Section 7.2: Preparation of the animal for surgery).

Virkon®-S (DuPont)

- Active ingredient: potassium peroxymonosulfate, 20.4%
- Controls bacteria, fungi and many viruses (including some activity against parvoviruses)
- Continues to work in presence of organic matter (unlike bleach)
- Dissolve tablets per manufacturer instructions (2 tablets per litre water).
- Use only on inanimate surfaces: floors, counter tops, tables, walls, cages surfaces.
- Do not use for instruments, endotracheal tubes, feeding tubes or other items that contact an animal's mucosa or internal structures. Do not apply to the animal directly.

VIROCID® or CID20® (www.cidlines.com)

- Composed of a quaternary ammonium compound, glutaraldehyde, and isopropanol.
- Dilute per manufacturer instructions.
- Appropriate for Quarantine or Isolation Unit (inanimate surfaces only).
- Use only on inanimate surfaces: floors, counter tops, tables, walls, cages surfaces.
- Do not use for instruments, endotracheal tubes, feeding tubes, food containers, water dishes or other items that contact an animal's mucosa or internal structures. Do not apply to the animal directly.

For further information on disinfection protocols and products, refer to www.vetmed.ucdavis.edu/msmp/protocols.htm. Manufacturer's instructions should be followed regarding the application and removal of residue of a particular cleaning product.

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Appendix 16: Legal forms for IFAW project participants

(following pages)

1. Volunteer registration form. Includes points of agreement to adhere to respectful and safe conduct.
2. IFAW Liability Release and Assumption of Risk form
3. Consent to surgery for spay / neuter
4. Consent to euthanasia of animal

Volunteer Registration Form

Volunteer Information		
Name:	Address:	City/State/Zip:
Home phone:	Work phone:	Cell phone:
Email address:		

Emergency Contact Information	
Name:	Relationship:
Home phone:	Alternate phone:

Medical Information		
Do you have medical insurance? ❑ Yes ❑ No Note: IFAW is not responsible for covering any medical costs for volunteers.	Carrier:	Policy number:
Do you have a current tetanus vaccination? ❑ Yes (date) ______________________________ ❑ No **Note: You must have a current tetanus vaccination. Proof of vaccination is required to volunteer.**		
Do you have a current rabies pre-exposure vaccination? ❑ Yes (date) ______________________________ ❑ No **Note: Rabies pre-exposure vaccination is recommended. By signing this form you are aware of this recommendation.**		
Are you on long term medications? ❑ Yes **Note: It is recommended that you inform your team leader of any long term medications you are on.** ❑ No		
Do you have any medical conditions that IFAW should be aware of in the event of an emergency? ❑ Yes (explain) ____ ❑ No		
Do you have any allergies? ❑ Yes (list)____________________________ ❑ No	Are you allergic to any animals? ❑ Yes (list)_____________ ❑ No	Are you afraid of any animals? ❑ Yes (list)_____________ ❑ No

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As a volunteer, I agree to the following:

- To represent IFAW in a professional manner;
- To follow the rules and procedures set up by IFAW for this project, including all veterinary protocols and emergency procedures;
- To respect IFAW's right to terminate me as a volunteer should it be determined that I am in conflict with the goals of the Organization to help animals during the project or if I am perceived to be a deterrent or threat to other volunteers and their well-being;
- To return to IFAW any property belonging to the Organization upon request;
- To use equipment and facilities belonging to, or being used by, IFAW in a manner not to damage or destroy them. Volunteers are responsible for replacing and/or repairing any property they intentionally damage or destroy;
- To not represent IFAW to the media without approval;
- I give IFAW permission to use videos and photographs taken during the course of the project and other activities that include images of me for use in broadcast videos, educational programs, newsletters, and advertisements. I understand I will receive no compensation in any form;
- To not abuse or neglect any animal;
- To not cause bodily harm to any other volunteers, IFAW staff, or other individuals cooperating with the project;
- To not bring weapons of any type on deployment;
- Illegal drugs are not permitted to be used at any time when volunteering for IFAW.

I have read the above mentioned conditions and agree to abide by them while volunteering for IFAW.

Volunteer signature: ____ Date: ______________________

IFAW LIABILITY RELEASE AND ASSUMPTION OF RISK

NOTICE: Participation in animal rescue and welfare projects can be a potentially dangerous activity. This Agreement prevents the bringing of a claim against IFAW or the IFAW Group (as defined below) in the event of death, illness or injury.

As a condition to, and in consideration of, being permitted to participate with a project (hereafter referred to as IFAW Project) associated with the International Fund for Animal Welfare Inc. (hereafter referred to as IFAW), the undersigned represents, acknowledges and agrees as follows, all for the benefit of IFAW and its affiliates in all forums, and their respective shareholders, members, directors, trustees, officers, employees, agents, representatives, insurers, successors and assigns (collectively, the "IFAW Group").

Fitness to Participate

I represent and verify that I am at least eighteen (18) years old, in good health, and aware of no physical problem or condition that will impair my ability to participate in the IFAW Project activities, including emergency response. I acknowledge and understand that IFAW is NOT in any way responsible for determining whether I am fit to participate in this program. I further represent that I will not use or possess drugs (including prescription medications which could impair my ability to participate in the IFAW Project), alcoholic, controlled substances or firearms while participating in the IFAW Project.

I understand that, prior to participating in the IFAW Project, it is recommended that I have a complete medical examination with a qualified physician, including the administration of such vaccinations for Hepatitis B, Tetanus and Rabies and other medical treatment as such qualified physician may deem necessary or advisable prior to or subsequent to the handling of animals.

Volunteer Status

I understand that, as a volunteer, I shall not receive any compensation whatsoever (including wages, salaries, benefits, unemployment insurance, workers compensation insurance or medical insurance) for my participation in the IFAW Project.

Assumption of Risk

I understand that participation in the IFAW Project involves a high degree of risk of injury to my person and/or death and injury to and destruction of my property. These risks may include, but are not limited to: strenuous physical activity and contact with animals that can be violent, carry disease and otherwise cause injury. I am voluntarily participating in the IFAW Project, with full knowledge and appreciation of this risk, and I understand and duly accept the potentially inherent dangers associated with the IFAW Project.

Medical Treatment

I give IFAW the right, in its sole discretion, but without any obligation to do so, to seek, authorize or approve medical treatment on my behalf, in the event that I am unable to so; and I agree to pay or reimburse IFAW for all costs associated with such medical treatment. No member of the IFAW Group shall be liable for the failure to seek or approve such medical treatment or for any damage, loss or injury resulting from any medical treatment provided on my behalf.

Insurance

I understand that IFAW does not carry or maintain health, medical, disability or life insurance coverage for any participant. Each participant is expected and encouraged to obtain his or her own health, medical, disability or life insurance coverage.

Release of Liability

On behalf of myself and my family members, companions, dependents, executors, administrators, heirs, assigns and representatives (collectively, the "Releasors"), I hereby release, discharge and hold harmless all members of the IFAW Group from all damages, losses, injuries, liabilities, claims, demands and causes of action for personal injury, death or damage to personal property ("Claims"), in each case suffered by me, by any Releasor or by any other person, arising from or occurring in connection with my participation or the participation of any other party or person in the IFAW Project, including, without limitation, injury, death or damage caused in whole or in part by the negligence or wrongdoing of any member of the IFAW Group, and any injury, death or damage arising out of any medical treatment or first aid provided or procured by IFAW. I agree that neither I nor any of other Releasor will ever assert in any forum any such Claim, and I shall indemnify and hold harmless all members of the IFAW Group from and against any such Claim (including reasonable attorneys' fees and costs incurred in defending such a Claim of any nature) brought against them by me or any Releasor.

Applicable Law; Jurisdiction; etc.

This IFAW Liability Release and Assumption of Risk is governed by the substantive laws of the Commonwealth of Massachusetts as if executed and to be formed in Massachusetts, without regard to conflict of law principles. I agree that all disputes arising under, in connection with or incident to, or related in any way to the IFAW Project or to this IFAW Liability Release and Assumption of Risk shall be litigated, if at all, in a state or federal court sitting in Massachusetts and I consent to the exclusive jurisdiction of such courts and waive, to the fullest extent permitted by law, any objection to the laying of the venue or that any such dispute has been brought in an inconvenient forum. If any provision contained in this IFAW Liability Release and Assumption of Risk shall be deemed invalid or unenforceable in whole or in part, this IFAW Liability Release and Assumption of Risk shall be enforced to the fullest extent allowed by law.

Miscellaneous

I represent to IFAW that I have advised my family members, companions and dependents of my participation in the IFAW Project, of the risks associated with such participation, and that I have assumed all such risks and released IFAW from any liability pertaining to such risks.

This document contains all of the agreements of the parties with respect to the subject matter hereof and supersedes all prior dealings of any nature between them with respect to such subject matter.

I have read and understand this IFAW Liability Release and Assumption of Risk and I am executing this IFAW Liability Release and Assumption of Risk voluntarily, without coercion and without reliance on any representation, expressed or implied, by any member of the IFAW Group. I understand that this IFAW Liability Release and Assumption of Risk waives important legal rights. I have had an adequate opportunity to consider this IFAW Liability Release and Assumption of Risk and to obtain such legal or other advice in regard to it as I considered advisable.

VOLUNTEER:

Full Name (Printed) ______________________________

Street Address of Legal Residence ______________________________

City, State, and Zip Code ______________________ Phone ______________

Signature/Date ______________________________

WITNESS:

Full Name (Printed) ______________________________

Street Address of Legal Residence ______________________________

City, State, and Zip Code ______________________ Phone ______________

Signature/Date ______________________________

Surgery consent form

Animal name: ____________________ Identification: ____________________

Species: ____________________ Breed: ____________________ Sex:______

Birth date: ____________________ ❑ estimated ❑ actual

Description of animal: ____________________

Surgical procedure(s) to be performed: ____________________

I give consent for the procedure/s listed above to be performed and understand that there is a slight element of risk involved with the administration of the anesthesia and the surgical procedure. I understand that the best possible care and techniques will be used to prevent any problems. In the event of any problems, I will not hold IFAW responsible in any way.

Furthermore, I verify that:

1. I understand that the veterinarian will clip some fur to make the site clean for the procedures. This fur will grow back within a few weeks following the surgery.
2. I understand that the veterinarian may not perform the procedure if he/she believes that the surgery will be an unreasonable risk to the animal's health.
3. I understand that following spay or neuter surgery, my dog/cat will not be able to reproduce.
4. I understand the instructions for taking care of my pet following surgery.
5. My animal last ate ______ hours ago and has had free access to water.
6. I have disclosed all information about my pet's health and medical history to the veterinary staff.
7. If my pet requires post-operative care by a veterinary professional, it is my responsibility to seek the appropriate care and to pay for it.

Printed name of owner/guardian: ____________________

Signature of owner/guardian: ____________________

Date: ____________________ Telephone no. ____________________

Witness: ____________________ Date: ____________________

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Consent for euthanasia

Animal name: ____________________ Identification: ____________________

Species: ____________________ Breed: ____________________ Sex:______

Birth date: ____________________ ❑ estimated ❑ actual

Description of animal: ____________________

Guardian:____________________ Tel:____________________

Reason for euthanasia: ____________________

By signing below, I agree for the veterinarian to euthanize the animal listed at the top of this form on the basis of reasons that I understand and with which I agree. The animal will be euthanized in a gentle and humane manner.

Printed name: ____________________

Relationship to animal: owner____________ rescuer____________ guardian____________ other ____________________

Signature: ____________________

Date: ____________________

Witness: ____________________ Date: ____________________

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GUIA DE CONTROLE HUMANITÁRIO DA POPULAÇÃO CANINA

Aliança Internacional para Controle de Animais de Companhia

RSPCA International

HUMANE SOCIETY INTERNATIONAL

IFAW

World Small Animal Veterinary Association

ARC

WSPA World Society for the Protection of Animals

Conteúdo

COVER IMAGE: ROBIN CULLEY/RSPCA PHOTOLIBRARY

Introdução

ICAM

A Aliança Internacional para Controle de Animais de Companhia (Aliança ICAM – International Companion Animal Management Coalition) é composta por representantes da Sociedade Mundial de Proteção Animal (World Society for the Protection of Animals - WSPA), da Sociedade Humanitária Internacional (Humane Society International - HSI), do Fundo Internacional para o Bem-estar Animal (International Fund for Animal Welfare - IFAW), da RSPCA Internacional (RSPCA International; braço internacional da Sociedade Real para Prevenção de Crueldade a Animais - Royal Society for the Prevention of Cruelty to Animals), da Federação das Universidades para o Bem-estar Animal (Universities Federation for Animal Welfare - UFAW), da Associação Mundial de Veterinários de Animais de Companhia (World Small Animal Veterinary Association - WSAVA) e da Aliança para Controle da Raiva (Alliance for Rabies Control - ARC).

Este grupo foi organizado para cumprir com diversos objetivos, incluindo o compartilhamento de informações e idéias a respeito da dinâmica populacional de animais de companhia, com o propósito de coordenar e melhorar as recomendações e orientações dos membros da organização. Cada organização concordou que é importante esforçar-se para melhorar nosso entendimento mútuo pela colaboração. Nós temos a responsabilidade, como financiadores e conselheiros, de garantir orientações acuradas, baseando-se nos mais recentes dados e conceitos disponíveis, para aqueles envolvidos com o controle populacional de cães em campo. Também acreditamos que é importante que nos esforcemos para sermos transparentes e que documentemos nossas opiniões e filosofias sempre que possível. É com este propósito que este documento foi produzido – ele representa nossas recomendações no momento em que é escrito, baseando-se no conhecimento que nós apuramos até o momento, e será revisado no momento apropriado. Nós temos plena ciência da falta de dados neste campo e vamos nos esforçar para fornecer novos dados e incorporá-los em nossas discussões, avaliações e diretrizes.

Novembro, 2007.

Para quem se aplica este guia

Este documento foi criado com o propósito de servir a órgãos governamentais e organizações não-governamentais (ONGs) envolvidos no controle populacional de cães.

A Aliança ICAM acredita que o controle apropriado da população canina é de responsabilidade do governo nos âmbitos local e federal. ONGs de bem-estar animal não devem ser encorajadas, tampouco procurar assumir a responsabilidade das autoridades governamentais sobre o controle populacional de cães a não ser por acordo contratual que inclua fundos e reservas apropriadas. Todavia, ONGs de bem-estar animal têm importante papel para conduzir e apoiar estratégias governamentais, portanto é importante que tais organizações entendam tudo que engloba uma estratégia abrangente. Isto irá possibilitar que foquem seu apoio onde poderá ser mais eficiente e para otimizar os limitados recursos existentes.

Propósito

Como defensora do bem-estar animal, a Aliança ICAM acredita que quando se julga o controle populacional de cães uma necessidade, é essencial que seja alcançado com emprego de práticas humanitárias e que por fim leve a uma melhora do bem-estar da população de cães como um todo. Como ONGs, nós também acreditamos que é importante que o controle da população seja alcançado tão eficazmente quanto possível devido às limitações dos recursos e também devido a nossa responsabilidade para com os doadores.

Este documento tem o propósito de orientar como avaliar as necessidades do controle populacional de cães e como decidir quanto à abordagem mais efetiva e eficiente (com relação aos recursos financeiros) deste controle com práticas humanitárias[1].

Nós também estamos cientes de que as condições, composição e tamanho das populações de cães podem variar significativamente entre países e dentro de um mesmo país, portanto não existe uma única intervenção que seja viável para todas as situações. Assim, nós defendemos fortemente a necessidade de uma avaliação inicial e a consideração de todos os fatores potenciais relevantes antes de decidir quanto à finalidade programada. O único conceito que consideramos universal é o da necessidade de um programa abrangente, que esteja focado nas causas e não somente no tratamento de sintomas da população de cães de rua.

1 Todavia em formato diferente e utilizando exemplos mais recentes, este documento compartilha muitos dos conceitos, especialmente com relação à avaliação inicial, incluídos no WHO/WSPA (1990) Guidelines for Dog Population Management.

Introdução

Todas as organizações dentro da Aliança ICAM têm como propósito comum e prioridade melhorar o bem-estar animal. O controle da população canina é uma área de interesse de todos nós porque incorre em questões de bem-estar.

Cães de rua podem ter uma série de problemas que afetam o seu bem-estar, incluindo:

- Desnutrição
- Doenças
- Ferimentos devido a acidentes de trânsito
- Ferimentos por brigas
- Maus-tratos.

Tentativas de controle da população podem também afetar significativamente o bem-estar animal, incluindo:

- Métodos desumanos de extermínio, como por envenenamento com estricnina, eletrocussão e afogamento
- Métodos cruéis de captura
- Locais de apreensão mal equipados e mal gerenciados

Dentro de cada população de cães existirão diferentes categorias. São estas:

- Com proprietário e mantidos com circulação restrita
- Com proprietário e com permissão para andar nas ruas
- Sem dono.

RSPA International

Cão que possui proprietário circulando nas ruas de Portugal.

Haverá questões de bem-estar relacionadas tanto aos cães mantidos com circulação restrita quanto aos de rua. No entanto, para os propósitos deste documento, o objetivo do controle da população canina é definido como: “Controlar a população de cães de rua e os riscos que podem representar, incluindo a redução da população quando for considerado for necessário”.

Apesar da redução do tamanho, até certo ponto, da população de cães de rua ser considerado um mal necessário, é subjetivo. Em cada situação existirão pessoas tolerantes, em relação aos cães de rua, outras não. Por exemplo, alguns membros da comunidade e autoridades governamentais estão preocupados com problemas de saúde pública e segurança associados aos cães de rua, incluindo:

- Transmissão de doenças para humanos (zoonoses) e outros animais
- Ferimentos e medo causados por comportamento agressivo
- Transtornos causados por barulho e sujeira
- Ataques e morte de gado
- Acidentes de trânsito

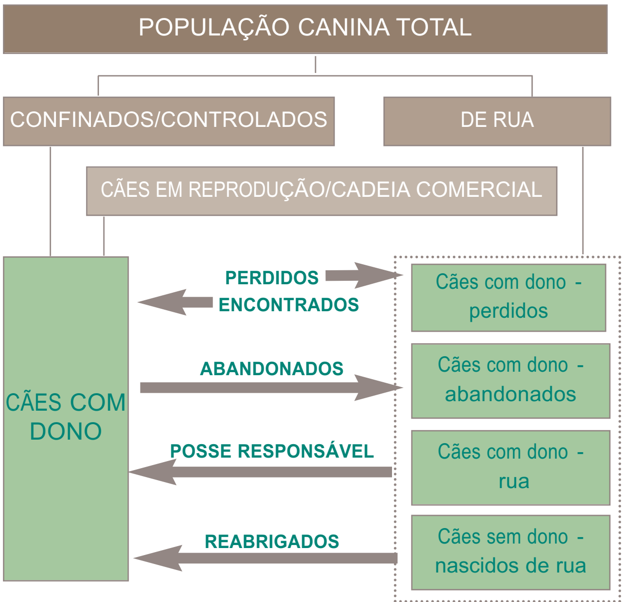
Por outro lado, em alguns países, cães que têm dono têm permissão para vagar sem restrições pela comunidade local. Uma redução do número de animais pode não ser necessária e tampouco almejada pela comunidade, todavia a melhora do bem-estar, da saúde da população e a redução dos riscos de zoonoses podem ser benéficos e desejáveis.

Um cão de rua pode ter dono ou não. É a guarda responsável de um cão que previne que seja considerado um problema por outros membros da comunidade. Este documento aborda opções de controle condizentes com as duas categorias, cães com dono e sem dono.

Terminologia

A partir de uma perspectiva de controle populacional, nós sentimos que é preferivel caracterizar os cães primeiro em relação ao comportamento e à área de circulação (em outras palavras, se vivem confinados ou nas ruas) e então pela categoria com ou sem dono. Isso está ilustrado na Figura 1, abaixo:

Figura 1: Sub-populações da população total de cães
O diagrama mostra as sub-populações nas quais a população total de cães pode ser dividida. Perceba que estas categorias são flexíveis e que cães podem se mover entre as categorias, como indicado pelas setas.



RSPCA International

Pescador e cão em comunidade na Índia.

Definições

Cão de rua

Aquele que não está sob controle direto ou que não está restrito por uma barreira física. Este termo abrange todos os animais de rua, tanto os que têm dono ou quanto os que não têm, sem distinguir se o cão tem um “proprietário” ou “guardião”; de fato, em muitos países, a maioria dos cães que seriam definidos como de rua na verdade têm um dono, todavia esses cães são deixados livres para andar por locais públicos a maior parte do dia.

Cão que tem dono

Para os propósitos deste documento, um cão que tem dono é aquele sobre o qual uma pessoa confere propriedade ou reivindica algum direito sobre – de maneira simplificada, quando perguntada sobre o cão, a pessoa irá dizer: “Este cachorro é meu”. Isto não significa necessariamente que seja um cão que tenha um dono responsável. Posse ou guarda sobre um cão pode variar de: posse “aberta” quando o animal de rua é alimentado sem regularidade nas ruas; até um cão mantido num estabelecimento comercial; até um cão (de estimação) bem cuidado, legalmente registrado e confinado. Na verdade, o que constitui a propriedade ou guarda de um cão é altamente variável e se enquadra em uma escala de confinamento, provisão de recursos como alimento e abrigo, e o significado da sua companhia.

Cão da comunidade

Podem ocorrer situações em que mais de um indivíduo reivindica posse de um animal e estes são conhecidos como cães da comunidade.

Posse ou guarda responsável de animais

É um princípio do bem-estar animal em que proprietários têm o dever de fornecer cuidado suficiente e apropriado a todos os seus animais e a seus descendentes. Este “dever de cuidar” exige que os proprietários ofereçam recursos (exemplo: comida, água, cuidados com saúde e interação social) necessários para um cão manter um nível aceitável de saúde e bem-estar em seu ambiente de convívio – as Cinco Liberdades[2] são um guia útil. Os proprietários também têm o dever de minimizar o risco potencial que seus cães podem representar para o público ou para outros animais. Em alguns países este é um requerimento legal.

2. Livre de fome e sede; livre de desconforto; livre de dor, ferimento ou doença; livre para expressar comportamento normal; livre de medo e perigo. Farm Animal Welfare Council (FAWC): www.fawc.org.uk/freedoms.htm

Estrutura do conteúdo

Este documento segue a estrutura explicada na Figura 2: Uma visão geral do processo, abaixo.

Figura 2: Uma visão geral do processo

E. Avaliação (página 19) Um processo periódico para reavaliar situação e estratégia

A. Coleta inicial de dados e avaliação (página 7)

- Qual o tamanho atual da população canina e de suas sub-populações?
- De onde vêm os cães e por que existe esta fonte?
- Quais os problemas causados pelos cães (real ou percebido) e o que está sendo feito atualmente para controlar estes problemas? Quem é o responsável por este controle?
- Quais problemas de bem-estar que os cães enfrentam?
- O que está sendo feito atualmente para controlar o tamanho da população e por quê? Quem é responsável?
- Quais são as partes envolvidas relevantes?

B. Fatores que influenciam o controle da população canina (páginas 8 - 11)

Considere e priorize fatores que afetem o tamanho da população:

- Caráter humano e comportamento
- Capacidade reprodutiva dos cães
- Acesso a recursos

Fatores que motivam realizar o controle animal:

- Zoonoses
- Atual população de cães de rua

C. Componentes de um programa abrangente de controle da população canina (páginas 12 - 16)

- Educação
- Legislação
- Registro e identificação
- Castração e contracepção
- Centros de assistência e de adoção
- Eutanásia
- Vacinação e tratamento
- Acesso a recursos controlado

D. Planejamento da intervenção (páginas 17-18)

- Planejamento para sustentabilidade
- Propósitos, objetivos e ações
- Estabelecimento de critérios para bem-estar animal
- Identificação de indicadores que podem ser usados para monitorar e avaliar cada etapa do programa

E. Implementação (página 19)

- Conduzir ações
- Manter mínimos padrões ao longo do processo

E. Monitoramento (página 19)

Um processo contínuo resultando em reajuste da implementação

A. Coleta inicial de dados e avaliação: Entenda o problema

Antes de iniciar um programa de controle da população canina, é essencial que a dinâmica da população de cães seja compreendida e mensurada objetivamente. Este caminho garante que o guia de controle final seja moldado às características da população de cães local, em vez de utilizar uma única dinâmica de intervenção para todos os cães ou situações.

Avaliação da população local de cães

As principais questões a serem exploradas através da avaliação são as seguintes:

1. Qual o tamanho atual da população canina e suas categorias intrínsecas? Isto inclui ambos os cães que tem dono ou não, confinados ou de rua, e onde estes se justapõem.
2. De onde vêm os cães de rua? Qual a fonte desses cães e por que esta fonte existe? Estratégias de controle devem procurar reduzir a população futura de cães de rua com alvo nas fontes primárias.
3. Quais as principais questões de bem-estar acercer desses cães?
4. O que está sendo feito atualmente, tanto informal quanto oficialmente, para controlar esta população de cães e por quê?

   a. Entender o que já está sendo feito pode permitir que fontes atuais e medidas de controle melhorem e aumentem. Isto também ajuda a garantir que quaisquer novas intervenções não serão conflitantes com as medidas correntes, porém as substituam ou as complementem.

   b. De quem é a responsabilidade de controlar a população de cães de rua? Geralmente essa responsabilidade é do departamento de agricultura (ou, por vezes, de saúde), com os municípios responsáveis por aplicar as ações localmente. ONGs podem fornecer elementos efetivos de controle populacional mas, no entanto, no intuito de fazê-lo, devem ter apoio em parceria ou serem liderados pelas autoridades responsáveis. Também é essencial que quaisquer medidas tomadas caibam na estrutura legal do país.

   c. Pressão popular pode ser bastante influente e este é geralmente o "porquê" por trás das tentativas de controle. É necessário ouvir as questões e opiniões da população e de autoridades locais; chamar a atenção para estas garantirá a sustentabilidade do projeto. A justificativa para se controlar as populações caninas irá depender das opiniões se os cães de rua são ou não desejáveis, mas nós sabemos que isto será determinado tanto pela pessoa que você está questionando quanto pelos cães individualmente em questão.

Para cada uma dessas questões estão sub-questões e ferramentas que podem ser utilizadas para tratá-las. Veja no Anexo A onde essas questões são analisadas, todavia note que as sub-questões e ferramentas descritas não são listas exaustivas ou obrigatórias, mas uma tentativa para chamar a atenção a áreas-chave de importância.

É essencial que todos os participantes relevantes sejam consultados durante este processo; deve ser procurada a representação de todos que sejam afetados pela população de cães. Tanto quanto possível, deve-se utilizar uma aproximação participativa; não somente as pessoas devem ser consultadas, como suas opiniões devem ser levadas em consideração e esta contribuição utilizada para planejar e direcionar intervenções futuras. Isto irá encorajar a participação dos envolvidos e irá, inevitavelmente, melhorar o sucesso do programa.

Criação de um comitê de participantes

O ideal é que a obrigação seja da autoridade governamental responsável trazer os envolvidos para uma consulta. No entanto, se eles não tiverem interesse ou viabilidade para fazê-lo, as ONGs podem criar um grupo de trabalho e repassar os resultados às autoridades relevantes. Para maiores informações sobre como desenvolver um processo consultivo, veja o Anexo B.

A seguir, uma lista dos possíveis envolvidos a serem consultados. Aqueles marcados com * são recomendados como requerimento mínimo do comitê.

Governo* - geralmente local, todavia o governo central é relevante para política e estatutos. Será o participante-chave se o programa tiver âmbito nacional. Diversos departamentos podem ser relevantes, incluindo de agricultura/veterinária, saúde, meio ambiente (especialmente no que diz respeito à coleta de lixo), turismo, educação e sanitário. O governo deve estar representado no comitê.

Comunidade veterinária* - conselho federal, associação profissional de veterinários, grupos privados de clínicos e departamentos de veterinária de universidades.

Comunidade de ONGs* - organizações locais, nacionais e internacionais que trabalhem com bem-estar animal, direitos dos animais e saúde humana.

Abrigos e ONGs que trabalham com adoção, lares temporários e realocação de animais* - regidos por governo/município ou organizações privadas/ONGs.

Comunidades acadêmicas com experiência relevante. Ex. comportamento animal, medicina veterinária, sociologia, ecologia e epidemiologia.

Legisladores* - departamentos responsáveis tanto por redigir quanto por fazer cumprir as leis.

Educadores – em escolas e universidades.

Mídia local – para educação, publicidade e apoio local.

Instituições internacionais com responsabilidades relevantes – Organização Mundial da Saúde (World Health Organisation – WHO), Organização Mundial de Saúde Animal (World Organisation for Animal Health – OIE) e associações veterinárias mundiais.

Líderes/representantes da comunidade local*.

Comunidade local – proprietários de cães ou não.

B. Fatores que influenciam o controle da população canina:
Considere uma gama de fatores que influenciam o bem-estar e o tamanho da população canina e decida qual priorizar

A avaliação inicial completa proverá tanto dados quanto critérios sobre a situação local. O próximo passo é realçar quais fatores são mais importantes e que devem ser priorizados no programa de controle; identificando estes fatores prioritários será garantido que os recursos não sejam gastos em questões que tenham impacto menor com relação ao problema maior. Em quase todas as situações, mais de um fator será importante, portanto uma estratégia efetiva irá requerer uma combinação de intervenções.

Abaixo, uma lista de fatores que são freqüentemente apontados como prioridades no controle da população de cães. Esses itens estão divididos naqueles que influenciam o tamanho da população e aqueles que influenciam ou motivam as pessoas na tentativa de controlar a população. No entanto, outros fatores podem ser relevantes em certas condições e é importante focar no que é apropriado para a comunidade alvo e as razões para a existência das populações dos cães de rua, e não apenas os efeitos.

Fatores que influenciam o tamanho da população canina

Comportamento e atitudes humanas

Objetivo: encorajar a guarda responsável.

O comportamento humano possivelmente é a força mais poderosa por trás da dinâmica da população canina. O encorajamento das interações homem-animal de maneira responsável e compensatória levará à melhora do bem-estar animal e a uma redução de muitas das origens de cães de rua. A população de cães que têm dono pode ser uma fonte significativa de cães de rua, podendo sofrer problemas de bem-estar que podem ser prevenidos. O comportamento humano em relação aos cães é a força propulsora por trás desses problemas.

Diversas questões devem ser consideradas ao se analisar as atitudes e o comportamento humano.

a. Crenças e hábitos locais podem afetar o comportamento humano relativo aos cães. É possível direcionar essas crenças para alterar atitudes. Por exemplo, a crença de que a cirurgia de esterilização causará alterações de comportamento negativas nos cães pode ser modificada através de educação e exemplos de cães esterilizados na comunidade, encorajando assim os proprietários a esterilizarem seus cães.

b. Manter uma comunicação coerente com o comportamento humano. A intervenção deve encorajar interações homem-animal responsáveis e compensatórias. Por exemplo, ao demonstrar tratamento respeitoso e cuidadoso para com os cães ajudará a encorajar posturas de empatia e respeito pela população local. É preciso ter atenção a quaisquer elementos da intervenção que possam ser vistos como comportamento irresponsável ou descuidado.

c. Religião e cultura têm papel importante nas crenças e hábitos das pessoas. Convide representantes religiosos e líderes da comunidade no início do processo, para entender como diferentes interpretações religiosas e culturais podem sustentar ou, pelo contrário, impedir intervenções.

d. Intervenções para alteração do comportamento humano devem ser moldadas cuidadosamente de acordo com o público-alvo, pois diferentes metodologias serão necessárias para diferentes idades e culturas. É importante conhecer os meios mais efetivos de comunicação para cada público alvo.

e. O comportamento humano é um fator chave para o sucesso do programa. Por isso, é importante que os proprietários não somente tomem conhecimento das intervenções, como também as compreendam amplamente e se engajem em todos os seus aspectos relevantes (veja o Estudo de Caso 1).

ESTUDO DE CASO 1

Exemplo de atitudes humanas que podem comprometer o controle da população canina

Na China, IFAW e One Voice financiaram uma pesquisa MORI em 2004. Esta revelou que aproximadamente 75 por cento dos cidadãos consideraram a esterilização cirúrgica de cães e gatos de estimação um ato de crueldade. Assim, ficou evidente a necessidade de uma campanha educativa e de uma discussão ampla antes de iniciar qualquer intervenção envolvendo o controle reprodutivo por esterilização de animais de estimação. Em 2006, houve uma situação similar em Zanzibar quando a WSPA e o governo local introduziram a esterilização cirúrgica. Começou com a baixa adesão de proprietários, sendo que poucos tinham a intenção de trazer seus animais para esterilização. No entanto, no período de um mês, como resultado do programa educacional, das discussões com líderes-chave da comunidade e dos exemplos reais de animais esterilizados e saudáveis, começou a haver mudanças no comportamento humano, até culminar com as pessoas trazendo espontaneamente seus animais para serem esterilizados.

WSPA

População local assistindo a cirurgia de esterilização através da janela de clínica móvel em Zanzibar.

Capacidade reprodutiva da população canina

Objetivo: equilibrar "oferta e demanda" para que o número e os tipos de cães nascidos estejam em conformidade com o número de cães requeridos pelo público.

Para diminuir o tamanho da população indesejada de cães de rua de modo humanitário é geralmente necessário reduzir o "excedente" populacional. Este excedente pode ser de cães com dono, sem dono ou que se reproduziram deliberadamente, e todas essas três categorias devem ser analisadas para o controle de oferta e demanda.

Os itens a seguir devem ser considerados.

a. Reduzir a reprodução. A esterilização pode reduzir a capacidade de reprodução, mas é importante selecionar com cuidado a população-alvo de cães.

i. Cães que estão se reproduzindo com sucesso.

- Para reduzir a taxa de reprodução da população com maior eficácia é importante avaliar quais cães estão efetivamente produzindo filhotes, os quais sobrevivem com sucesso até a idade adulta.
- Alguns estudos de populações específicas de cães que não estavam recebendo cuidados diretos de humanos (ex. estavam vivendo de restos encontrados em lixeiras) indicaram que o tamanho da população fora mantido devido à imigração contínua de outros animais para o grupo, ao invés do acasalamento com sucesso dos animais do grupo. Isto posto, pode-se dizer que em muitos casos somente aqueles animais recebendo algum cuidado direto de pessoas terão mais sucesso reprodutivo.
- Através da perspectiva do bem-estar animal, deve se considerar que os filhotes nascidos de cadelas que vivem em condições precárias sofrem (isto quando estas cadelas são capazes de manter a gestação até o nascimento dos filhotes). Em geral, é provável que a taxa de mortalidade dos filhotes seja alta nas populações de cães de rua e sem dono.
- Cães que estejam em condições de bem-estar desfavoráveis podem posteriormente melhorar o estado de saúde e tornarem-se aptos a se reproduzir com sucesso.

ii. Cães cujos filhotes estão sujeitos a se tornar de rua.
Pode haver populações específicas de cães cujos filhotes estão mais sujeitos a se tornarem animais de rua ou a serem abandonados. Isto pode estar relacionado com a falta de conhecimento e aceitação dos princípios da guarda responsável, que pode ser resultado de educação, iniciativas públicas e institucionais, e sócio-econômicas.

iii. Fêmeas (cadelas). Pode ser razoável concentrar esforços para intervir junto às fêmeas, que geralmente são o fator limitante da capacidade reprodutiva de uma população de cães. São necessários apenas alguns machos férteis (não-esterilizados) para fertilizar fêmeas férteis no cio. Deste modo, castrar uma proporção maior de machos pode não reduzir a capacidade reprodutiva total da população. Cada fêmea castrada, todavia, contribuirá individualmente para a redução da capacidade reprodutiva da população no todo.

iv. Cães machos. O comportamento sexual dos machos não-esterilizados pode ser problemático, especialmente quando as fêmeas não-esterilizadas estão no cio. É possível que os machos adultos não alterem seu comportamento de maneira significativa após a esterilização, diferentemente dos machos jovens e que ainda não desenvolveram seu comportamento sexual. Conseqüentemente, o grupo dos machos jovens pode ser a próxima prioridade para esterilização, seguido dos machos adultos.

Nota: *Tanto machos quanto fêmeas podem ser vetores da raiva. Assim se somente as fêmeas estão sendo selecionadas para castração em área endêmica de raiva, os machos devem ao menos ser vacinados.*

b. Reduzir a fonte comercial, como a procriação de cães para venda. Uma estratégia abrangente deve ponderar as fontes comerciais de cães, como os canis para reprodução e os *pet shops*. Estabelecimentos comerciais de reprodução de cães podem produzir filhotes pouco sociáveis e doentes, considerados de baixa qualidade. Lojas, como mercados e pet shops, também podem ter animais em condições desfavoráveis e podem vendê-los sem orientação adequada quanto a cuidados ou a responsabilidades. A "baixa qualidade" desses cães e a falta de entendimento ou de expectativas realistas sobre a guarda desses animais pelos proprietários levarão a um alto risco de abandono. O cumprimento das leis e a fiscalização por agentes especializados podem melhorar as condições desses estabelecimentos comerciais e assim o bem-estar dos animais envolvidos. As lojas também devem dar orientação adequada quanto aos cuidados e às responsabilidades da guarda de cães. Pode -se instruir prováveis proprietários de cães para que conheçam as opções disponíveis ao adquirirem um novo animal de estimação, incluindo os centros de realocação de animais. Assim, essas pessoas podem buscar um filhote socializável e saudável.

Acesso a recursos

Objetivo: reduzir o acesso dos cães a recursos que possam encorajá-los a andar pelas ruas à procura disso e direcionar os recursos locais para reduzir a população local de cães de rua.

Cães geralmente têm acesso a recursos (como alimento, água e abrigo) quando fornecidos diretamente por seu proprietário em sua moradia ou quando encontrados em áreas públicas quando estão pelas ruas. Quanto um cão precisa dos recursos disponíveis em áreas públicas para sua sobrevivência, isto depende do nível de cuidados proporcionados pelo dono. Alguns cães que têm dono são encorajados a andar nas ruas porque têm acesso a recursos em áreas públicas, mas não dependem destes para sobreviver. Já outros cães de rua que não têm dono ou não são cuidados pelo dono são, portanto, totalmente dependentes desses recursos para sobreviver. Alterar o acesso aos recursos em áreas públicas trará um impacto na população de cães de rua, pois irá desestimular os passeios oportunos de cães com dono. Contudo, também poderá reduzir potencialmente a sobrevivência daqueles que dependem destes recursos.

Diversas questões devem ser consideradas quando se investiga este fator.

a. A intervenção para redução do acesso a recursos não deve ser utilizada isoladamente. Sobre os animais identificados como dependentes de recursos disponibilizados em áreas públicas para sua sobrevivência, as alterações no acesso a tais recursos (através de medidas como melhora do sistema de coleta de lixo) devem ser feitas juntamente com a redução da população ou elaborando provisões alternativas para esses animais.
b. A melhora do sistema de coleta de lixo e conseqüente descarte podem reduzir o contato entre pessoas, especialmente crianças, e cães de rua.
c. Em algumas situações, a principal fonte de alimento será aquela fornecida por pessoas, em vez daquela encontrada no lixo (abastecimento indireto de recursos). Os motivos que levam as pessoas a alimentar os cães de rua irão variar entre as áreas geográficas e entre os indivíduos, e isso deve ser entendido e levado em consideração quando se tenta influenciar este comportamento; como exemplo, veja o item d, abaixo. Educar as pessoas será uma ação importante na tentativa de influenciar este comportamento. Todavia, a redução da população de cães pode levar automaticamente a uma redução dos recursos disponibilizados pelas pessoas, pois estas não terão mais cães para alimentar.
d. A alteração do acesso aos recursos em áreas específicas pode ser utilizada para alterar a distribuição da população canina. Por exemplo, para manter um determinado parque público livre de cães de rua circulando, pode-se remover o acesso aos recursos, com utilização de lixeiras a prova de animais e educando as pessoas para não alimentarem os cães nessas áreas. Em alguns países existem regras restritivas, que limitam as áreas onde os cães podem se exercitar ou caminhar livremente. Essas regras são impostas por fiscais públicos e de meio ambiente.

Fatores que motivam pessoas para o controle da população canina

Zoonoses (doenças que podem ser transmitidas dos animais para o homem)

Objetivo: reduzir o risco que a população de cães representa para a saúde humana e para a saúde de outros animais.

As zoonoses são muitas vezes a causa primária de preocupação em relação às populações de cães de rua, particularmente para os governos local e federal, que têm responsabilidade quanto às questões de saúde pública. A raiva é uma doença fatal e os cães são os vetores de transmissão mais comuns para humanos, por isso o controle dessa doença é freqüentemente o principal motivo para controle da população canina.

Diversas questões precisam ser analisadas quando se investiga este fator.

a. A importância do controle de zoonoses não deve ser minimizada pelos participantes relevantes, como os órgãos oficiais de saúde pública. É importante pesquisar em conjunto os caminhos para o controle efetivo das zoonoses, que devem ser neutros, ou até mesmo positivos, em relação ao bem-estar animal.
b. As zoonoses são de interesse da população em geral e as pessoas podem por vezes agir cruelmente contra cães, ainda que estes não sejam uma ameaça para a transmissão de zoonoses, como a raiva. Controlar as zoonoses e fornecer evidências tangíveis desse controle (ex. colocar colares vermelhos para indicar os animais que foram vacinados recentemente) para que a população veja pode ajudar a aumentar a confiança e a reduzir o comportamento agressivo contra esses cães.
c. Em algumas situações é aconselhável introduzir um programa de controle de zoonoses melhorado para, primeiramente, reconquistar a confiança da população, e então seguir com outros elementos de controle populacional de cães, como esterilização ou melhoria dos cuidados com a saúde. Todavia, um programa abrangente de controle populacional, incluindo o controle de zoonoses, é a opção ideal.

Vida Digna

Cães de rua alimentando-se de lixo no Peru.

d. Deve-se considerar o risco de transmissão de zoonoses para as pessoas envolvidas em qualquer intervenção de controle populacional. Por exemplo, cães com raiva podem excretar o vírus na saliva por até duas semanas antes do aparecimento dos sintomas. Todos os envolvidos que trabalhem em contato com cães devem receber treinamento, equipamentos adequados e medicação profilática (preventiva).

População atual de cães de rua

Objetivo: reduzir os riscos que a população de cães de rua apresenta para a comunidade e evitar a redução de seu bem-estar.

A população de cães de rua pode levar a conflitos ente homem e animal (acrescido das zoonoses) e se tornar um problema de bem-estar evidente. Em muitas situações a população de cães de rua deverá ser controlada devido à pressão da população sobre questões de saúde pública e de bem-estar dos próprios animais. O melhor método de se controlar esta população dependerá muito da comunidade local de pessoas e da própria população de cães.

Diversas questões devem ser analisadas quando se investiga este fator.

a. É importante identificar exatamente onde e por que ocorrem os conflitos entre homem e animal. Pode ser possível resolver alguns conflitos através de outros métodos que não aqueles que tenham por objetivo reduzir a população, como instrução para prevenção de mordidas ou estabelecimento de zonas livres de cães em áreas de possíveis conflitos.

b. Os conflitos entre homem e animal e as questões de bem-estar são geralmente as principais razões da origem de uma população de cães de rua sem dono, já que na verdade muitos desses cães de rua podem ter sido abandonados pelo seu antigo dono. A melhora da guarda responsável de animais e a introdução de registro e identificação de cães são métodos para se tentar resolver esta questão. Maiores detalhes são fornecidos na Seção C.

c. Pode haver lares temporários na comunidade local que poderiam abrigar com responsabilidade cães de rua sem dono. Para administrar esta questão, um centro de realocação ou um sistema de lares temporários seriam necessários, embora ambos necessitem de controle criterioso para não causarem problemas para si próprios. Centros de realocação podem ser caros e demandam tempo para administrar, portanto é melhor explorar alternativas criativas antes de se comprometer com um espaço físico. Veja a Seção C para maior detalhamento destas questões.

d. Em alguns casos não haverá ou haverá poucos centros potenciais de realocação. Nesta situação, o bem-estar dos cães deverá ser levado em consideração. Em muitos casos, o bem-estar limitado desses cães e a pressão pública significarão que estes animais precisam ser removidos. Se estiverem doentes, machucados ou com problemas de comportamento significativos, como agressividade, a eutanásia poderá ser a melhor opção. Se não existem lares disponíveis, a eutanásia poderá ser preferível, por questões de bem-estar animal, ao invés de mantê-los em abrigos por longos períodos de tempo, visto que é difícil e caro abrigar cães por tanto, tempo sem sofrimento significativo.

e. Se o bem-estar desses cães é geralmente bom e a comunidade local os tolera, é possível introduzir uma combinação de medidas para controlá-los *in situ*, incluindo: vacinação da população de cães para garantir que não sejam transmissores da raiva; utilização de uma “ambulância” para recolher os animais que estiverem machucados, doentes ou que sejam agressivos, para eutanásia humanitária; manutenção de zonas livres de cães através do recolhimento e do bom isolamento do lixo. Estas medidas devem ser utilizadas em conjunto com outras desenvolvidas para administrar as fontes de origem desta população. Maiores detalhes na Seção C.

f. A matança indiscriminada de cães utilizando-se métodos desumanos é, infelizmente, freqüentemente utilizada como uma tentativa de controlar a população. Há muitas razões para que isto não ocorra. Eliminar cães de rua não atinge a fonte de origem dos animais e, portanto, esta medida deverá ser repetida indefinidamente. Este método encontra resistência freqüentemente, tanto local quanto perifericamente, posto que o tratamento desumano de um animal vivo será visto como eticamente questionável, especialmente quando existem alternativas humanitárias disponíveis. Se os métodos desumanos utilizados são também indiscriminados, tais como a utilização das iscas com veneno, também haverá o risco para outras espécies, outros animais de estimação e até mesmo para humanos. Não há evidência que sugira que o extermínio em massa reduza a incidência da raiva (veja o Estudo de Caso 2) e pode, na verdade, desencorajar os proprietários de cães a se engajar em programas de prevenção da raiva, quando estes são conduzidos por autoridades que são conhecidas por selecionar indiscriminadamente.

Há indícios de que em alguns casos o extermínio em massa leve à redistribuição dos animais sobreviventes em novos territórios. Isso pode na verdade aumentar o risco de raiva por aumento do movimento das populações. Também é uma hipótese de que em uma situação onde a procriação é limitada por acesso a recursos, uma rápida redução dos animais por extermínio em massa pode permitir maior acesso a recursos para os animais remanescentes. Dessa forma, seu sucesso reprodutivo e sobrevivência aumentariam potencialmente, possibilitando que eles rapidamente repusessem os animais selecionados. Todavia, até o momento não temos conhecimento de dados que demonstrem estes efeitos.

ESTUDO DE CASO 2

Um exemplo da ineficácia do extermínio em massa de cães para o controle da raiva

Flores é uma ilha isolada na Indonésia que fora livre de raiva até que um surto de raiva canina resultou em pelo menos 113 mortes de pessoas. O surto começou depois que três cães foram importados de Sulawesi, região endêmica de raiva, em setembro de 1997. Autoridades locais responderam com extermínio em massa de cães, no início de 1998. Aproximadamente 70 por cento dos cães do distrito onde a raiva havia sido introduzida foram mortos naquele ano e, no entanto, a raiva canina ainda existia em Flores até o momento da publicação do estudo (junho de 2004).

De Windiyaningsih et al (2004). *The Rabies Epidemic on Flores Island, Indonesia (1998–2003)*. Journal of the Medical Association of Thailand, 87(11), 1-5.

C. Componentes de um programa abrangente de controle da população canina: Selecione as soluções mais apropriadas para a sua situação

Um programa eficaz de controle da população canina necessita de uma abordagem abrangente. O ideal é que o programa geral seja coordenado pela autoridade local responsável pelo controle populacional de cães. ONGs devem trabalhar com a autoridade para identificar as áreas em que eles podem dar assistência e maior contribuição ao programa. Todas as ações devem ser selecionadas baseando-se nas prioridades identificadas na avaliação das necessidades iniciais. Esta seção esboça uma variedade de componentes que podem se tornar parte de um programa abrangente de controle populacional de cães.

Educação

A longo prazo, a educação das pessoas é um dos elementos mais importantes de uma abordagem abrangente para o controle populacional de cães, assim como o comportamento humano é um fator influente extremo na dinâmica da população canina (veja Seção B). Em geral, a instrução educativa precisa encorajar uma maior responsabilidade, entre os proprietários, com relação ao controle populacional de cães, aos cuidados mínimos e ao bem-estar individual dos animais. Contudo, podem existir mensagens específicas relacionadas às instruções que são importantes para salientar as diferentes etapas do programa, como por exemplo: prevenção de mordeduras; seleção e cuidados com os cães; expectativas realistas sobre a guarda de cães; divulgação da importância dos tratamentos preventivos e do acesso aos mesmos; conhecimento dos comportamentos caninos normais e anormais.

Diversas questões devem ser analisadas quando se investiga este fator.

a. Iniciativas educacionais devem ser desenvolvidas em conjunto com as autoridades educacionais locais e realizadas por profissionais treinados. Todos os participantes podem recomendar conteúdo e impulsionar os programas, mas a prática deve ser feita com apoio profissional.

b. É importante conectar todas as fontes possíveis de educação a respeito de cães para garantir que as mensagens sejam consistentes. O ideal é incluir grupos de bem-estar animal, veterinários, escolas, grupos de fiscalização e mídia (incluindo grupos com foco em animais). Pode ser necessário que um grupo em particular coordene o programa.

ESTUDO DE CASO 3

Exemplo de um programa educacional

Após o tsunami de 2004, o Blue Paw Trust desenvolveu um programa educacional em conjunto com uma clínica veterinária móvel nas costas sul e leste do Sri Lanka. Foram distribuídos panfletos sobre cuidados com cães e gatos e palestras foram ministradas em centros comunitários e escolas locais, havendo discussões na própria clínica entre os membros da equipe veterinária e o público. No final, os proprietários de animais de estimação foram apresentados aos veterinários locais que freqüentaram a clínica em apoio ao programa e também para conhecer técnicas estéreis de cirurgia. Essas iniciativas educacionais foram planejadas e moldadas com contribuição de escolas e autoridades locais (agentes de saúde pública) e ocorreram em sincronia com outros grupos de bem-estar animal locais.

c. Poderá ser necessário focar os esforços na instrução de veterinários e estudantes de veterinária na área de controle populacional, incluindo:

- Os princípios que conduzem, ou as justificativas para o controle populacional
- O papel dos veterinários nas questões de saúde pública
- Métodos de controle reprodutivo
- Mensagens-chave para clientes sobre guarda responsável
- Métodos de eutanásia
- Como eles podem se envolver e se beneficiar de programas pró-ativos de controle populacional que estimulem o cuidado responsável de cães, incluindo o cuidado veterinário habitual.

d. Mensagens educacionais podem ser transmitidas de diversas maneiras, incluindo:

- Seminários e aulas em escolas
- Panfletos para o público-alvo
- Desenvolvimento da consciência no público geral através da imprensa, cartazes, rádio e televisão
- Envolvimento direto de pessoas nas discussões como parte dos programas-base da comunidade (veja o Estudo de Caso 3).

e. O impacto da educação no controle da população de cães pode levar tempo para se tornar evidente, portanto métodos de monitoramento e avaliação deste impacto devem ser incorporados a indicadores, tanto de curto como de longo prazo. Este impacto pode ser compreendido em três diferentes aspectos: desenvolvimento de conhecimento e habilidades; mudanças de atitudes; alteração do comportamento resultante.

Legislação

É essencial que o programa de controle populacional de cães esteja de acordo com os códigos de leis – e que preferencialmente tenha o apoio delas. A legislação é importante para a sustentabilidade do programa e pode ser utilizada para assegurar que o controle populacional de cães seja feito dentro dos padrões humanitários. A legislação relevante pode estar tanto nos níveis locais quanto federais, e algumas vezes é diversificada entre diversos estatutos, leis ou decretos diferentes. Demais documentos legais também podem ser relevantes e podem ter impacto na importância ou no método de aplicação das leis. Alterar a legislação pode ser um processo longo e burocrático.

Diversas questões devem ser analisadas quando se investiga este fator.

a. Há um equilíbrio a ser alcançado entre a legislação que é clara e aquela que de tão restritiva, não permite evoluir nas práticas de controle populacional.
b. Deve-se despender tempo para planejar cuidadosamente uma nova legislação, delineando-a a partir de experiências relevantes de outros países e profissionais. Deve ser utilizado um processo inclusivo com a participação de todos os envolvidos relevantes, incluindo exercícios de avaliação, procurando a contribuição ativa e incorporada de diferentes fontes.
c. É difícil alterar a legislação vigente, portanto é importante que as propostas para novas versões de leis sejam precisas e realistas. Finalmente, as novas leis devem ser: holísticas; consideradas cabíveis e razoáveis para a comunidade; que envolvam as autoridades com suas responsabilidades; que alcancem o impacto desejado para o bem-estar animal; que sejam sustentáveis.
d. Deve haver tempo suficiente para que sejam absorvidas quaisquer alterações da nova legislação. Deve-se fornecer com antecedência guias de utilização das novas leis para auxiliar na interpretação das mesmas.
e. As leis ficarão apenas “no papel” a menos que sejam ratificadas consistentemente e impostas eficientemente. Para a ratificação efetiva, geralmente a maior parte do esforço é com instrução e incentivos e a menor parte gasta em medidas punitivas. Deve-se almejar ensinar sobre a nova legislação em todos os níveis, desde os órgãos que aplicam a lei (como advogados, polícia e inspetores de bem-estar animal) a profissionais relacionados (como veterinários e gerentes de abrigos) e proprietários de cães. A aplicação da lei tem sido vitoriosa em alguns países através da criação de cargos como os de inspetores de bem-estar animal (também conhecidos como guardiões ou oficiais de controle animal). Estes oficiais são treinados e capacitados para instruir, manipular animais quando requerido e reforçar as leis com conselhos, avisos, cuidados e eventuais ações legais.

Registro e identificação

O modo mais efetivo de vincular claramente o proprietário com seu animal é utilizando, concomitantemente, métodos de registro e identificação. Isto deve instigar um senso de responsabilidade no proprietário, pois o animal capturado poderá ser identificado. Registro/identificação é uma ferramenta importante para devolver os animais perdidos aos seus respectivos proprietários e pode ser um importante recurso para aplicação da legislação (incluindo leis de abandono e vacinação anti-rábica compulsória).

Diversas análises podem ser feitas quando se investigam estas questões.

a. Existem diversos métodos disponíveis de identificação de animais, e estes podem ser utilizados tanto isoladamente quanto em conjunto. Esses métodos diferem de três importantes maneiras: quanto à permanência, à visibilidade e se o animal deve ou não ser anestesiado quando são aplicados os métodos de identificação. *Microchips*, tatuagens e colares são os três métodos mais comuns; o mais aplicável dependerá em parte das condições do local e em parte das razões pela qual a identificação está sendo utilizada.
b. Se for necessária a identificação permanente de uma grande população, atualmente o microchip é a melhor opção. O número de permutações de dígitos do código é suficiente para identificar todos os cães, e erros humanos (por transposição e leitura incorreta de números) ocorrerão com menos freqüência, pois um scanner digital é utilizado na leitura do chip. A utilização de microchips também tem a vantagem de ser uma tecnologia difundida globalmente, assim os animais que se deslocam de uma área (ou país) para outro podem ainda ser identificados (veja Estudo de Caso 4). Antes de instituir um sistema de microchipagem, é prudente checar se os chips e os leitores estão em conformidade com os padrões ISO.
c. É importante que as informações de registro e identificação sejam guardadas em um banco central de dados (ou que bancos de dados estejam em conexão de alguma maneira), que sejam acessíveis para todas as pessoas relevantes (ex. veterinários, polícia, guardiões de cães e abrigos municipais). Pode ser necessário o apoio do governo federal para garantir a utilização de um sistema unificado.
d. Registro e identificação compulsórios podem ajudar nos problemas reais enfrentados pelos abrigos. Quando um cão trazido para um abrigo é identificado, pode ser devolvido sem demora para o seu proprietário (evitando o comprometimento do bem-estar do cão e reduzindo o estresse do proprietário). Se não pode ser identificado, é por definição “sem dono”, portanto o abrigo pode aplicar seus procedimentos (adoção ou eutanásia) sem esperar o proprietário se apresentar. Em ambas as circunstâncias, haverá mais espaço nos canis, o que potencialmente aumentará a capacidade dos mesmos.

ESTUDO DE CASO 4

Exemplo de um sistema de identificação e registro na Estônia

O governo da cidade de Tallinn foi o primeiro a adotar um sistema de registro e identificação compulsório para cães na Estônia. O sistema foi introduzido em agosto de 2006 como um projeto piloto, quando a cidade de Tallinn financiou uma empresa para desenvolver um arquivo de dados para gravar e identificar animais e seus proprietários.

Leis municipais estipulam que todos os cães devem ser identificados permanentemente por um *microchip* que fora implantado por um veterinário. Os detalhes dos proprietários e de seus animais são gravados num arquivo de dados que pode ser acessado por pessoal autorizado. O registro foi desenvolvido para ser universal, permitindo que o mesmo sistema seja utilizado em toda Estônia. O sistema será utilizado tanto para identificar os animais quanto para eventualmente emitir chamados de vacinação compulsória anual contra raiva.

e. Podem ser cobradas taxas de registro (pagamento único ou anual) no intuito de prover fundos para outras áreas do programa de controle populacional. Porém, recomenda-se atentar para equilibrar os valores das taxas de registro com as das multas a serem aplicadas pela não utilização do registro, já que taxas muito altas podem levar os proprietários a evitar o registro, não se importando com o pagamento das multas. Diferentes escalas de valores de taxas podem ser utilizadas como um incentivo para a castração dos cães, encorajando proprietários a manter somente um pequeno número de animais e desestimulando a procriação de seus cães.
f. Sugere-se que critérios sejam adotados para a aquisição de cães com a emissão de licenças para diminuir os problemas, por exemplo, de procriação sem controle e de aquisição de animais de raças controladas (cães "perigosos"). As licenças podem encorajar a guarda responsável, pois solicitam que os novos proprietários completem um "certificado de posse de cão" antes que lhes seja concedida a licença para possuir um.

Esterilização e contracepção

O controle reprodutivo por esterilização ou contracepção temporária pode ser conseguido através de três métodos principais.

a. Cirúrgico: a remoção de órgãos reprodutores sob anestesia geral garante a esterilização permanente e pode reduzir significativamente o comportamento sexual (principalmente se realizado nos estágios iniciais de desenvolvimento de um animal). As técnicas cirúrgicas devem ser executadas corretamente. Deve-se manter um bom padrão de técnicas de assepsia (a prática para reduzir ou eliminar os riscos de contaminação bacteriana) e de controle da dor durante o procedimento, o que somente pode ser avaliado por monitoramento pós-cirúrgico durante todo o período de recuperação. A cirurgia pode parecer onerosa no início, mas é uma solução para toda a vida e se considerar o longo prazo, o custo é baixo. O procedimento requer veterinários com prática, infra-estrutura e equipamento adequados.
b. Esterilização química e contracepção: estes métodos são ainda um pouco limitados devido ao custo, à pela necessidade de repetição e aos problemas de bem-estar associados com o uso de alguns químicos. Atualmente, nenhum método de esterilização química ou de contracepção tem garantia de eficácia e nenhum descarta riscos à saúde quando utilizados em cães de rua não monitorados. No entanto, esta é uma área de pesquisa atual e esperam-se esterilizantes químicos eficazes e apropriados para controle reprodutivo em massa no futuro. Requer-se o exame clínico para a avaliação do estado reprodutivo dos indivíduos por veterinários experientes antes da aplicação da maioria dos químicos. Além disso as injeções devem ser administradas em intervalos regulares sem interrupção, condição não aplicável à maioria dos programas de controle de cães. Esterilizantes químicos e contraceptivos devem ser utilizados de acordo com as instruções dos fabricantes, que podem ou não ter impacto no comportamento sexual dos animais.
c. Contracepção física pelo isolamento das fêmeas durante o cio: os proprietários podem ser instruídos para reconhecer os sinais de uma fêmea entrando no cio e isolar a fêmea dos machos durante todo esse período. Deve-se atentar ao bem-estar tanto de fêmeas quanto de machos quando se planeja o isolamento das fêmeas. O comportamento sexual dos machos pode se tornar problemático ao procurar acesso às fêmeas, no entanto o isolamento tem custo mínimo e não requer um cirurgião veterinário com prática.

Ao utilizar ferramentas para castração e contracepção é importante considerar a sustentabilidade dos mesmos – o controle da população canina é um desafio constante, portanto é vital que se considere planejar uma intervenção sustentável. Fornecer serviços de graça ou a custo baixo, sem explicar os custos totais envolvidos, pode dar uma expectativa irreal do verdadeiro custo do cuidado veterinário aos proprietários de cães.

Uma infra-estrutura veterinária local é um requerimento para a saúde geral e o bem-estar de animais que tenham proprietários. Portanto, se uma clínica veterinária privada local pode oferecer serviços de esterilização, é recomendável trabalhar para melhorar e incorporar esta capacidade de prestação de serviços. Em vez de excluir ou alienar esta clínica do programa. Esta prática pode receber o apoio de um crescente "mercado" à procura de serviços de esterilização de cães na comunidade local, que acredita nos benefícios deste procedimento, ajuda a financiar parte dos custos e também apóia a melhora dos serviços veterinários através de treinamento (veja Estudo de Caso 5).

ESTUDO DE CASO 5

Exemplo de um programa para desenvolvimento de um controle populacional sustentável envolvendo participantes locais

Uma avaliação profunda da população canina local, que combinou uma pesquisa doméstica formal e a contagem dos cães conhecidos no local, forneceram informações sobre as fontes de cães de rua na República Dominicana e, portanto, a percepção do "problema". Como resultado, o comitê da cidade exigiu o controle dos cães na cidade, por força de lei, de maneira humanitária e efetiva. O comitê solicitou à IFAW a complementação do seu programa municipal através do fornecimento de cuidados veterinários primários (incluindo castração) e instrução, através de um programa de extensão porta a porta na comunidade, baseando-se nos resultados da avaliação. O objetivo era limitar o número de animais de rua na fonte, e também concentrar-se em questões de bem-estar de cães com dono, tais como negligência, confinamento inapropriado e saúde debilitada. A filosofia do programa almejava a participação e a liderança comunitária, assim os veterinários locais eram parte integral do projeto. Seguindo programas de treinamento, tanto na República Dominicana quando em outros países, funcionários do IFAW dos EUA e do Reino Unido forneceram apoio à distância a funcionários-chave locais e a demais envolvidos, assim como também forneceram protocolos veterinários por escrito cabíveis às condições locais e aceitáveis nos padrões internacionais. Através deste processo a comunidade local, os veterinários e o conselho poderão liderar todas as frentes do projeto a longo prazo.

Para discussão dos resultados do questionário à comunidade, veja Davis et al (2007), Preliminary Observations on the Characteristics of the Owned Dog Population in Roseau, Dominica. JAAWS

Abrigos e centros de realocação

A construção de abrigos não irá por si só solucionar a questão dos cães de rua a longo prazo. Pode até piorar o problema, pois é um caminho fácil para os proprietários de animais de estimação simplesmente abandoná-los ao invés de cuidar dos mesmos. Além disso, centros de realocação podem ser onerosos e demandam tempo para administrar, portanto alternativas criativas a estes centros devem ser exploradas antes de se começar a construir um. Um sistema de lares temporários, por exemplo, pode ser mais efetivo, mais eficiente e proporcionar melhores condições de bem-estar aos animais (veja Estudo de Caso 6). Ao invés de um centro de realocação, que trata os sintomas de abandono em vez das causas, os esforços deveriam ter foco prioritário na melhora da guarda responsável como um método de redução do abandono. Se já existem abrigos legais de animais de rua e observação dos casos de suspeita de raiva, como por exemplo, os abrigos gerenciados pelo município ou por fundações, pode ser menos dispendioso melhorar e expandir estes abrigos que a construção de novos.

ESTUDO DE CASO 6

Exemplo de uma alternativa aos centros de realocação

Em uma cidade ao leste da Ásia, com uma das maiores densidades populacionais humanas do mundo, uma grande população de cães de rua e, todavia, uma limitada capacidade para angariar fundos, muitos abrigos rapidamente se tornaram superlotados. Em muitos casos, a falta de recursos financeiros e a constante demanda levaram a uma queda dramática dos padrões de cuidados, resultando em sofrimento para os animais e aflição para os funcionários. Como alternativa, uma nova organização criou uma rede de fomento de voluntários, dedicados a levar cães e gatos abandonados para suas casas temporariamente. Para tanto, a organização concordou em dar assistência aos animais, pagando por todas as contas médicas, vacinas e esterilização, até que fossem encontrados lares definitivos para esses animais. No primeiro ano a organização construiu uma rede de mais de 40 lares temporários com o objetivo de atingir 100 no segundo ano. Os animais são realocados via internet e a rede tem possibilidade de alojar um número muito maior de animais que um abrigo jamais pôde. Todos os animais são mantidos em condições apropriadas e a organização tem muito menos despesas administrativas que um abrigo. A nova organização se tornou um sucesso em uma cidade onde muitos outros projetos similares falharam.

Adaptado de Guidelines for the design and management of animal shelters, RSPCA International, 2006. –151.

Diversas questões devem ser analisadas quando se investigam estes fatores.

a. Políticas de ação deverão ser redigidas para cobrir diversas questões de importância, incluindo esterilização, adoção, capacidade (quantos animais por canil e no total, e o que será feito se a capacidade for excedida) e eutanásia. Deve-se levar em consideração o bem-estar dos animais, as implicações com os custos, as metas e os objetivos do centro e o impacto deste na questão do controle populacional de cães a longo prazo, incluindo esterilização responsável. Como este é um tópico onde fatores emocionais estão envolvidos, é preferível que as políticas de ação sejam acordadas por todos os funcionários desde o início. Todos os novos funcionários devem estar cientes das políticas de ação e devem entender a lógica claramente.

   Exemplo 1: uma política de ação clara e seus procedimentos devem estar de acordo para que seja possível avaliar a saúde e o comportamento de cada cão, tendo em mente os lares que estarão disponíveis e o que se pode esperar realmente de um novo lar. A adoção inapropriada pode levar à desconfiança do público e significar relações públicas ruins para adoção em geral.

   Exemplo 2: dando continuidade ao Exemplo 1, alguns cães não serão apropriados para adoção por causa de sua saúde e/ou comportamento e pode não haver lares suficientes para aqueles que seriam apropriados. É extremamente difícil proporcionar boas condições de bem-estar para os cães ao mantê-los em canis por longos períodos de tempo. Nessa situação, deve-se considerar a eutanásia tanto para esses animais quanto para outros cães aos quais não poderiam ser oferecidas oportunidades de encontrar um novo lar. Para apoiar a tomada de decisão, as políticas de ação sobre a eutanásia devem ser claras e transparentes para todos os funcionários envolvidos.

b. Protocolos devem ser elaborados para cada estágio do processo, da quarentena na chegada à rotina diária que inclui limpeza, alimentação, exercício, manutenção da documentação e realocação.

c. Pode-se levar em consideração as necessidades para o bem-estar dos animais ao se planejar um centro, incluindo tanto necessidades fisiológicas quanto psicológicas. A escolha do local deve levar em consideração o acesso do público, as características físicas, as provisões (como drenagem e fornecimento de água), o possível barulho gerado, os alvarás e as expansões futuras.

d. As finanças dos centros de realocação são extremamente importantes, pois é difícil encerrar suas atividades de uma hora para outra. Deve-se levar em consideração os gastos de capital e os custos de gerenciamento. Recomenda-se que ambos sejam acumulados para o período de um ano antes de se comprometer com um novo centro de adoção.

Para mais informações, consulte: *Guidelines for the design and management of animal shelters*, RSPCA International, 2006.

3. Ex. veja definições fornecidas por Asilomar Accords: http://www.asilomaraccords.org/definitions.html

Eutanásia

Quando abrigos, centros de realocação e suas redes estão operando, faz-se eutanásia dos animais que estejam sofrendo de doenças incuráveis, ferimentos ou problemas de comportamento que os impeçam de ser realocados, ou que não estejam se adaptando às condições do local para que mantenham um nível mínimo de bem-estar. Enfim, um programa de controle populacional de sucesso deve criar uma condição onde estas sejam as únicas situações em que a eutanásia é necessária e que os animais saudáveis possam encontrar um bom lar. Na verdade, a maioria dos países não atingirá esta situação de imediato, mas será necessário trabalhar para isto, aceitando que alguns animais saudáveis terão de ser sacrificados enquanto não existirem lares que possam oferecer um bom nível de bem-estar.

A eutanásia soluciona apenas os sintomas e não as causas dos problemas populacionais. A eutanásia por si só não é um método de controle populacional e não deve ser a única responsável por esse controle. Quando se utilizar a eutanásia, devem ser utilizados métodos humanitários para que o animal fique inconsciente antes que sua morte ocorra, sem sofrimento.

Vacinação e controle parasitário

Podem ser oferecidos tratamentos veterinários preventivos para proteger a saúde e o bem-estar dos animais e reduzir o problema das zoonoses. As vacinações anti-rábicas são geralmente questões prioritárias, mas pode haver vacinas para diversas outras doenças e, concomitantemente, o controle parasitário interno e externo, que deve ser feito com medicação apropriada. Esses tratamentos devem ser fornecidos em conjunto com instruções sobre guarda responsável, esterilização e contracepção, e registro e/ou identificação. Geralmente os proprietários compreendem a necessidade das vacinas e do controle parasitário e assim o acesso a tais serviços pode ser o modo mais fácil para atrair essas pessoas para conversas e acordos sobre outros fatores discutidos nesta seção.

Diversas questões devem ser analisadas quando se investigam estes fatores.

a. Vacinação periódica (especialmente se compreender outras doenças que não só a raiva) e controle parasitário podem melhorar a qualidade da saúde dos cães. Fêmeas que antes não tinham capacidade reprodutiva podem melhorar a saúde a ponto de conseguirem novamente se reproduzir. Isso significa considerar a questão do aumento populacional minimizado-o como requerido.
b. Assim como a esterilização e a contracepção, podem ser utilizados tratamentos preventivos para influenciar os proprietários no reconhecimento do valor dos tratamentos veterinários em geral e de outras ferramentas de controle populacional (tais como registro e identificação), requeridos para o bem-estar dos animais a longo prazo. Portanto, vale a pena estudar como envolver a infra-estrutura veterinária local para fornecer tratamentos preventivos. Os tratamentos preventivos gratuitos devem ser feitos com atenção e de acordo com a situação econômica local, pois existe o risco da desvalorização dos serviços veterinários em geral se o tratamento for fornecido sem custo algum ou sem o entendimento da extensão dos custos subsidiados.
c. Tratamentos preventivos deverão ser feitos rotineiramente para terem eficácia, assim deve-se considerar facilitar o acesso aos tratamentos.

WSPA/ BLUE PAW TRUST

Colocação de colar de identificação vermelho em um cão que recebeu vacina anti-rábica e tratamento anti-parasitário no Sri Lanka.

d. Os tratamentos podem ser improvisados em "acampamentos" (em locais temporários, com alta concentração de animais), bastante eficazes para alertar os proprietários quanto à importância dos tratamentos preventivos e outras ferramentas de controle populacional. No entanto, deve ser minimizado o risco de interações agressivas e de transmissão de doenças entre o grande número de animais no local Isso pode ser possível com a organização cuidadosa dos acessos e das saídas do local, com a utilização de seringas esterilizadas para cada animal e com a colocação dos animais doentes em quarentena. Esses acampamentos devem ser divulgados amplamente com antecedência. Deve-se considerar a distância limite que o público em geral irá se deslocar para usufruir de tais serviços, portanto deve-se pensar sobre o número de acampamentos necessários para atender ao número de animais desejado e a logística associada.
e. Encorajar o tratamento preventivo regular permite o diagnóstico e a terapêutica de quaisquer condições existentes.

Controle de acesso aos recursos

Cães são encontrados em locais públicos onde existe acesso a recursos, como comida. Para restringir essa condição, especialmente em áreas onde os cães não são tolerados (ex. escolas e parques públicos), deve-se restringir o acesso a recursos. Isto deve ser feito com cuidado e em conjunto com demais medidas de redução da população de cães de rua, a fim de evitar que os cães fiquem famintos quando as fontes de alimento são removidas ou que migrem para outras áreas em busca de novas fontes.

Isto pode ser conseguido de diversas maneiras:

a. removendo regularmente o lixo das casas e das lixeiras públicas
b. cercando os locais de coleta e de depósito de lixo
c. controlando o destino das sobras e descarte de carcaças
d. utilizando lixeiras que dificultem o acesso dos animais, como aquelas com tampas pesadas, ou colocando-as longe do alcance dos cães
e. instrução e medidas de coação para impedir que as pessoas despejem o lixo em locais inadequados (e dessa maneira alimentarem os cães acidentalmente) e prevenir que as pessoas alimentem propositadamente os cães em determinadas áreas.

D. Planejamento da intervenção: Preparação, acerto dos objetivos e ajuste dos padrões

Uma vez que a avaliação esteja completa, que as prioridades para o programa tenham sido decididas e que a abordagens para tentar resolver estas prioridades tenham sido exploradas, é necessário esboçar e documentar tod o plano do programa.

Planejamento para sustentabilidade

Os programas de controle populacional de cães geralmente requerem muitos recursos por um longo período de tempo, incluindo recursos humanos, infra-estrutura e finanças. É importante considerar os seguintes fatores.

a. Responsabilidade: o ideal é que a solicitação de recursos à autoridade responsável seja feita de acordo com as finanças disponíveis. É mais provável que os órgãos públicos alcancem a sustentabilidade através do financiamento do próprio governo. ONGs que queiram se responsabilizar por determinados aspectos do controle da população canina devem se certificar dos recursos disponíveis, quer das autoridades, quer de outras fontes, antes de assumir tais responsabilidades. Estas ONGs também devem considerar que seu comprometimento será de longa duração, o que poderá desafiar sua capacidade de assumir outra frente de trabalho.

b. Envolvimento dos proprietários: uma intervenção projetada para ter um impacto sobre a guarda responsável pode levar à sustentabilidade de elementos do projeto, como também a alterações de comportamento positivas e permanentes. Por exemplo, programas de castração poderiam se tornar sustentáveis se os proprietários fossem encorajados a pagar pelos serviços, sendo parte desses serviços veterinários subsidiados para que os preços ficassem mais acessíveis.

c. Registro: um sistema de registro de posse de cão, cobrando-se uma pequena taxa, poderia fornecer recursos para outros componentes do programa. Todavia, o valor da taxa deve ser controlado cuidadosamente, pois taxas altas podem desestimular a adesão ao sistema de registro. A cobrança de taxas pode não ser apropriada em todos os países.

d. Obtenção de fundos: a capacidade para angariar recursos localmente dependerá de diversos fatores, incluindo a cultura de doação e o significado dos cães na comunidade local. Pessoas da localidade, negócios, empresas de crédito e indústrias relacionadas a cães (farmacêutica, de alimentos e de seguros) podem ter interesse em apoiar os programas de controle de cães, quer financeiramente, quer provendo recursos (como alimentos e remédios). Grupos internacionais para obtenção de recursos também podem custear projetos específicos, mas dificilmente irão se comprometer com projetos de longa duração. Novamente a sustentabilidade de cada uma das fontes de fundos e/ou recursos deve ser considerada.

e. Recursos humanos: pode haver pessoas interessadas em apoiar o programa oferecendo recursos humanos sem custo algum, o que por vezes é chamado de doação pro bono. Diversas profissões realizam trabalho pro bono em benefício de ONGs, como marketing, finanças e empresas de administração. A profissão veterinária é um importante recurso humano, não somente por suas qualificações em cirurgia e medicina, mas também pela habilidade dos veterinários em influenciar o comportamento de proprietários de animais. Veterinários podem querer oferecer algum tipo de serviço gratuitamente ou a custo reduzido. Estudantes de veterinária também podem querer auxiliar como parte de seu treinamento, que pode se tornar parte formal do curso de veterinária, todavia é necessário supervisionar os trabalhos. Veterinários voluntários e enfermeiros veterinários de outros países também podem ser fontes valiosas de apoio, todavia há a possibilidade de serem considerados uma ameaça aos veterinários locais se estes interpretarem que os demais vêm para substituir seus serviços. A sustentabilidade desta fonte também é difícil, pois os custos com as viagens podem ser altos. Pode ser preferível utilizar veterinários voluntários locais para apoiar o desenvolvimento e as habilidades da profissão veterinária.

f. Sustentabilidade: deve-se traçar um plano de sustentabilidade a longo prazo desde o início; o controle humanitário da população de cães tem um início mas não um fim, pois requer ação constante para manter determinada população na condição desejada. Inclusão e desenvolvimento de acordo com as capacidades locais apoiarão a sustentabilidade e também a guarda responsável, pois os proprietários apoiarão ações de controle populacional.

Propósitos, objetivos e ações

O plano do programa deve incluir os propósitos e os objetivos claros e acertados. Também é importante neste estágio descrever os indicadores que poderiam ser utilizados para avaliar o progresso em cada etapa do programa. Os indicadores serão utilizados para monitorar e avaliar o sucesso do programa (veja Seção E) e é importante considerá-los desde o início, pois os princípios básicos do programa poderão ser requisitados futuramente. Se existe um número considerável de organizações envolvidas no programa, pode ser relevante preparar acordos para que cada parte esteja ciente do objetivo a ser alcançado e do seu papel dentro do programa. Estes planos devem ser comunicados aos usuários finais, como proprietários de cães e envolvidos que serão afetados pelo programa, ainda que não sejam responsáveis pelas ações eles próprios (podem-se incluir determinadas autoridades). Veja Estudo de Caso 7 para um modelo de controle populacional de cães.

Estabelecimento de critérios para o bem-estar animal

O objetivo de manter o melhor nível possível de bem-estar animal deve estar claramente declarado nos princípios do programa. Para garantir conformidade e compreensão, os princípios são mais bem desenvolvidos quando criados por um grupo de participantes. As decisões relacionadas ao destino dos animais devem ser tomadas com base tanto em seu bem-estar a longo prazo como com relação à população de cães local. Deve haver um procedimento de monitoramento freqüente para garantir que estes princípios estão sendo endossados, e também de revisão dos próprios critérios.

A seguir, áreas dos programas de controle de cães que podem requerer o emprego de princípios mínimos:

a. cirurgia, incluindo técnicas de assepsia, anestésicos e administração de medicamentos (ex. analgesia)
b. manipulação e transporte de cães
c. alojamento e cuidados com cães
d. procedimentos de adoção
e. eutanásia – quando e de que maneira deve ser aplicada
f. manutenção da documentação e análise freqüente dos dados – apesar de não afetar diretamente o bem-estar animal, a boa organização de documentos que reúnam os casos de doenças ou ferimentos podem ajudar a identificar partes do programa que possam comprometer o bem-estar. Por exemplo, a alta incidência de complicações pós-operatórias em determinados períodos pode indicar a necessidade de treinamento para determinadas equipes veterinárias ou de mudança nos cuidados pós-operatórios.

RAY BUTCHER/PHUKET ANIMAL WELFARE SOCIETY

Cirurgia com técnicas de assepsia, Tailândia.

ESTUDO DE CASO 7

Exemplo de ações para o desenvolvimento de intervenções

A. Entenda a situação

Aplicou-se um questionário à população do município X, que teve o maior número de queixas contra cães de rua. As respostas ao questionário mostraram que 50 por cento das pessoas que tinham cadelas afirmaram que têm muitos filhotes para cuidar e que encontrar lares para esses é um problema. Também afirmaram que 45 por cento dos filhotes eram “perdidos”. A taxa de esterilização cirúrgica de cadelas era de apenas três por cento. Os proprietários comentaram que não tinham muita confiança na habilidade dos veterinários locais e havia uma preocupação de que seus cães tivessem a personalidade alterada como resultado da esterilização.

B. Priorize os fatores relevantes

O fator relevante neste caso é o da reprodução dos cães – existe um excedente de filhotes rejeitados dentre a população de cães com donos, uma necessidade de aumentar as taxas de esterilização dos cães com donos, e uma necessidade de concentrar-se na habilidade dos veterinários, e a falta de compreensão quanto ao impacto da esterilização sobre o comportamento canino.

C. Componentes de um programa abrangente

Os componentes são: esterilização cirúrgica através da infra-estrutura veterinária local; instrução tanto de veterinários para os procedimentos cirúrgicos quanto de proprietários sobre a importância da esterilização.

D. Desenvolva a intervenção

A partir deste ponto, descreveu-se o propósito: reduzir o número de cães indesejados e de cães de rua susceptíveis a doenças e ferimentos nas ruas do município X. Para alcançar este propósito, diversos objetivos foram traçados, um dos quais fora aumentar a esterilização de cadelas que têm dono de três para 50 por cento em dois anos. Este prazo fora escolhido devido à disponibilidade de recursos (tempo da clínica e apoio financeiro) e para dar tempo que o impacto da campanha se tornasse evidente.

Este objetivo envolverá ações, assim como:

- treinamento para melhorar as habilidades de esterilização cirúrgica em quatro clínicas veterinárias locais, o que está sendo amparado por dois incentivos: um sistema de cupons permitindo que veterinários ofereçam serviços de esterilização a custo baixo, subsidiados por uma ONG local, e um plano de marketing simples para a clínica sobre esterilização a custo baixo
- uma campanha educacional, utilizando anúncios e a rede comunitária local com foco no líder religioso, que explica aos proprietários os benefícios da esterilização de cães em relação à saúde e ao comportamento.

E. Implantação, monitoramento e avaliação: Verifique se o programa está atingindo os objetivos

Implantação

Deve ser simples se as prioridades foram escolhidas com prudência e o plano elaborado em detalhes. Este estágio pode requerer uma abordagem em fases, utilizando áreas-piloto que são cuidadosamente monitoradas para garantir que quaisquer problemas sejam resolvidos antes do lançamento da campanha. Os estágios iniciais não devem ser antecipados. Haverá problemas básicos e participantes-chave irão requerer informações freqüentes, para monitorar de perto e melhorar o progresso nos estágios iniciais.

Monitoramento e avaliação

Quando o programa estiver em andamento, será necessário monitorar freqüentemente o progresso e avaliar sua eficácia. Isto é necessário para:

a. ajudar a melhorar o desempenho, ao salientar tanto os problemas quanto os elementos de sucesso das intervenções

b. a confiabilidade, ao demonstrar aos doadores, aos apoiadores e às pessoas beneficiadas no final da intervenção que o programa está alcançando seus objetivos. O monitoramento é um processo contínuo para checar se o programa está advindo de acordo com o planejado e para fazer ajustes freqüentes. A avaliação é periódica, geralmente praticada quando ocorrem determinados eventos, para checar se o programa está tendo o impacto desejado e definido. A avaliação deve ser utilizada para embasar as decisões com relação a investimentos futuros e à continuidade do programa. Ambos os procedimentos envolvem a mensuração de indicadores selecionados no estágio de desenvolvimento porque refletem importantes componentes do programa nos diferentes estágios (veja Estudo de Caso 8 para exemplo).

O monitoramento e a avaliação devem ser partes importantes do programa, mas que não despendam muito tempo ou recursos financeiros. A escolha dos indicadores corretos com relação a suas capacidades de refletir mudanças, as quais podem ser mensuradas com certo grau de confiança, será a chave do sucesso neste estágio. Para a escolha destes indicadores é essencial ter um plano claro com relação ao que o programa se determina a alcançar e por que, e como, a intervenção atingirá isso.

O ideal é que o monitoramento e a avaliação sejam abordados de maneira participativa, onde todos os envolvidos relevantes sejamconsultados e chamados para fazerem recomendações. É também importante permanecer aberto às possibilidades e com atitude positiva durante este processo, pois podem ocorrer mudanças contrárias às expectativas.

A exposição dos problemas ou falhas deve fazer com que estes sejam vistos como oportunidades para melhorar a campanha, e não como erros a serem justificados.

Os conceitos de monitoramento e avaliação não são complexos, porém existem muitas decisões que devem ser tomadas com relação ao que mensurar, como isto pode ser feito e como os resultados podem ser analisados e utilizados. Estas questões e outras são discutidas em muito mais detalhes em outros textos. Para exemplo, visite: www.intrac.org.

ESTUDO DE CASO 8

Matriz de projeto sugerindo indicadores para cada fase do projeto inicialmente introduzido no estudo de caso 7

HIERARQUIA DE OBJETIVOS		**INDICADOR** *Medida, número, fato, opinião ou percepção que reflita uma condição específica ou situação*	**MEIOS DE VERIFICAÇÃO** *Como você medirá o indicador*
IMPACTO/META Reflete a mudança gerada pelo projeto	Redução de cães de rua indesejados no município X	Diminuição na % do número dos filhotes errantes e cadelas lactantes no município X após 2 anos	Censo populacional bianual (contagem direta em amostra de quarteirões de 500m²)
RESULTADO/PROPOSTA Reflete o efeito do projeto	Aumento da habilidade da comunidade em controlar a capacidade reprodutiva de seus cães	Porcentagem de fêmeas esterilizadas aumenta para 50% após 2 anos	Questionário anual para as residências
		Aumento na % da comunidade para aceitação da esterilização de cães	Grupos de discussão focando a comunidade
META 1 Reflete o esforço realizado pelo projeto	4 esquemas de esterilização a baixo custo no município X	Número de cães esterilizados e tratados por mês	Recorde de clínicas participantes
ATIVIDADES 1 Reflete o que o projeto realmente fará	1.1 treinamento para 4 veterinários locais 1.2 Desenvolver sistema de cupom 1.3 Marketing de serviços a baixo custo	Número de clínicas qualificadas e inscritas no esquema	Acordo com clinicas

ANEXO A: Ferramentas para avaliar as deficiências do controle populacional de cães

Este anexo tem o objetivo de explorar as questões que são comentadas na Seção A. Abaixo de cada cabeçalho está uma série de sub-questões e correspondentes sugestões de ferramentas que poderiam ser utilizadas para investigar tais questões. O objetivo não é de fornecer uma lista detalhada ou descritiva, porém de encorajar a exploração do assunto.

1. Estabelecer uma estimativa do tamanho da população canina e suas categorias

SUB-QUESTÕES	SUGESTÕES DE FERRAMENTAS/MÉTODOS
Quantos cães estão atualmente nas categorias "de rua" e "confinados"? Atenção, pois cães de rua serão tanto sem como com dono.	Um levantamento da população de cães de rua em conjunto com um questionario para os donos de cães locais perguntando sobre o número de cães que normalmente estariam à solta no momento em que a pesquisa era conduzida. Note que é necessária experiência durante a elaboração dos questionários para se obter dados confiáveis e relevantes.

2. Para entender de onde vêm os cães de rua. Em outras palavras, quais as fontes desses animais e por que existem estas fontes?

SUB-QUESTÕES	SUGESTÕES DE FERRAMENTAS/MÉTODOS
Com o tempo, como está se alterando e como está se mantendo a população de cães de rua? A população de cães sem dono é capaz de se reproduzir com sucesso por si só? Os cães sem dono têm condições de criar os filhotes até a idade adulta?	Observe o número de animais em cada classe de idade (filhote, jovem e adulto) da população de cães de rua ao longo do tempo. Observe o número de filhotes durante a estação de reprodução de cães de rua com e sem dono para verificar quantos sobrevivem nas duas populações.
Os cães indesejados com dono, abandonados nas ruas, se tornam parte da população dos cães de rua? Os cães com dono têm permissão para andar livremente?	Questionário para os proprietários – pergunte se seus animais estão confinados em propriedade particular ou se eles (ou outro alguém que eles saibam; se admitirem este comportamento eles próprios é possível que seja um problema) alguma vez abandonaram um cachorro.
Se o abandono ou os cães de rua são um problema, por que isto ocorre? Quais as crenças, atitudes e fatores ambientais que estão subordinados a estes comportamentos?	Atitudes e crenças por trás de tais comportamentos podem ser difíceis de mensurar confiavelmente (utilizando uma escala numérica habitual). Discussões ou entrevistas de modo aberto com grupos de pessoas com experiência relevante (como proprietários de cães ou pessoas que trabalhem com saúde animal) podem ajudar a discutir opiniões. Faça grupos pequenos e informais e permita discutir livremente os tópicos, utilizando perguntas instigantes para guiar a discussão.

IFAW/S.COOK

Levantamento junto a proprietários de animais de estimação na República Dominicana.

3. Quais os problemas de bem-estar enfrentados pela população canina e por que ocorrem?

A mensuração de bem-estar pode ser abordada por uma avaliação do animal (observação direta dos animais), por avaliação dos recursos (mensurando o acesso que os animais têm a recursos importantes para seu bem-estar) ou por uma combinação de ambas. Mensurar o bem-estar em populações de cães, especialmente aquelas que incluem uma proporção de animais de rua, é uma área relativamente pouco estudada. No entanto, é importante para nós, como defensores do bem-estar animal, que tentemos direcionar estas questões em algumas sub-questões importantes.

SUB-QUESTÕES	SUGESTÕES DE FERRAMENTAS/MÉTODOS
Qual o nível de bem-estar de uma população de cães de rua e como prevalecem os problemas de bem-estar?	Observação direta do nível de saúde de cães de rua, como escore corporal, claudicação, ferimentos e problemas de pele.
Qual o nível de bem-estar de cães com donos e como prevalecem os problemas de bem-estar? Os proprietários dão a seus cães os recursos necessários para o bem-estar de seus animais?	Observação direta de cães com donos para verificar o estado de saúde e a resposta comportamental ao dono (para descobrir o tratamento anterior do dono com relação ao seu cão). Questionar os donos em relação aos cuidados com saúde, alimento, água e abrigo.
Qual o nível de bem-estar dos cães atualmente afetados pelas medidas de controle? Por exemplo, qual o nível de bem-estar dos cães em abrigos? Quais métodos de eutanásia são utilizados, se é feita a eutanásia?	Observação direta dos cães em abrigos, utilizando os mesmos critérios para outras categorias de cães para haver comparação. Discussões com autoridades dos abrigos sobre as provisões fornecidas e métodos de eutanásia utilizados.
Quais as taxas de sobrevivência dos diferentes tipos de cães (confinados, sem dono ou de rua com dono) ou diferentes grupos de idade? Sobrevivência pode indicar nível de bem-estar e sobrevida curta poderia indicar saúde debilitada.	É difícil mensurar a sobrevivência das populações de rua sem dono sem utilizar uma amostragem de indivíduos ao longo do tempo. Um questionário para proprietários sobre cães em seus lares que morreram ao longo do ano anterior pode fornecer uma estimativa da sobrevivência de cães com dono e as razões porque estes animais morreram (observe que a sobrevivência de filhotes jovens e adultos deve ser levada em consideração separadamente, posto que estes são geralmente bastante diferentes).

4. O que está sendo feito atualmente para controle populacional de cães, tanto informal quanto oficialmente e por quê?

SUB-QUESTÕES	SUGESTÕES DE FERRAMENTAS/MÉTODOS
As pessoas pensam que existe um problema com o controle populacional de cães na localidade? Quais problemas são causados por estes cães?	Discussões com pequenos grupos de pessoas de diferentes origens. Faça grupos pequenos e informais e permita discutir livremente os tópicos, utilizando perguntas instigantes pertinentes para guiar a discussão. Pergunte às autoridades locais sobre a natureza, número e localização geográfica das queixas.
O que está sendo feito atualmente para controle da população canina?	Discussões com todos os envolvidos relevantes para compreender os planos passados, presentes e futuros para controle de população canina. Considere os governos locais, organizações veterinárias, ONGs e os próprios proprietários.
Qual a legislação vigente a respeito do controle populacional de cães?	Acumule informações tanto dos governos federal quanto local quanto à legislação relativa aos cães – é possível que leis pertinentes estejam descritas em mais de um Ato (ex. controle de doenças, regulamentos veterinários, regulamentos ambientais).

ANEXO B: Criação de um comitê de participantes

Abaixo, exemplo de um processo que pode ser utilizado para conseguir envolvimento de participantes; tal processo pode ser adaptado a diferentes tamanhos de programas (de pequenos projetos comunitários a programas nacionais).

- Criação de um grupo de trabalho com pessoas com interesse e responsabilidade quanto ao controle populacional de cães (veja Seção A para uma lista de possíveis envolvidos). Este grupo de trabalho terá a responsabilidade de planejar e coordenar a coleta de dados inicial e avaliar a população canina local.
- Depois de uma avaliação inicial, este grupo de trabalho pode estar envolvido com um comitê formal de representação de cada parte relevante. O comitê deve, no mínimo, ter termos de referência, uma lista de membros e regras para os mesmos, comprometimento com reuniões regulares, novidades sobre um plano de ação e objetivo definido. É possível basear este comitê em modelos similares, por exemplo, aqueles criados para melhora da saúde humana. Também é importante convidar membros experientes daqueles comitês para o de controle populacional de cães.
- Cada membro do comitê é responsável por representar as necessidades de seus envolvidos com relação ao controle populacional de cães. Por exemplo, os órgãos de saúde pública requerem controle de zoonoses, as ONGs requerem melhora de bem-estar e o conselho municipal requer redução das queixas contra distúrbios. Um conjunto de objetivos pode ser delineado, baseando-se nos dados produzidos pela avaliação inicial e pelas necessidades de cada parte. O plano do programa pode se formar ao redor deste, com entendimento claro dos objetivos e o que será visto como sucesso ou falha por cada parte (veja Seção D para mais informações de como montar um plano).
- O comprometimento financeiro requerido para uma campanha bem-sucedida, tanto a curto como a longo prazo, deve ser discutida e acordada pelo comitê. Isto deve incluir o investimento esperado por cada envolvido.
- A responsabilidade de cada membro do comitê em executar, monitorar e avaliar o programa deve estar clara. Uma vez que o programa é lançado, reuniões freqüentes serão necessárias para trazer informações recentes quanto ao progresso e à discussão dos resultados de monitoramento e avaliação e, portanto, quaisquer mudanças necessárias ao programa.
- O comitê será essencialmente permanente, posto que o controle populacional de cães é um desafio contínuo, todavia a participação será inevitavelmente alterada e evoluirá ao longo do tempo.

A seguir, sugestões para melhora do funcionamento do comitê.

- Seminários ou workshops podem ser utilizados para inspirar e desenvolver o programa em pontos chave, incluindo o estágio de planejamento. Este tipo de evento pode também trazer à tona conhecimentos geralmente não presentes no comitê.
- Esclarecer regras, incluindo detalhes de questões administrativas (ex. minutas e organização de reuniões), ajudará a criar expectativas realísticascondizentes com a realidade. As regras também devem ser freqüentemente revistas e revisadas, quando apropriado.
- Tanto quanto possível o comitê deve ser transparente, para encorajar a confiança do público no programa.
- O comitê irá vivenciar inevitavelmente diferenças de opiniões, portanto um programa claro e um entendimento de como tais situações serão gerenciadas ajudarão a manter coesão.

The Alliance for Rabies Control
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World Small Animal Veterinary Association
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