

BLUETONGUE (INFECTION WITH BLUETONGUE VIRUS)

SUMMARY

Bluetongue (BT) is an infectious, non-contagious, vector-borne viral disease that affects wild and domestic ruminants such as sheep, goats, cattle, buffaloes, deer, most species of African antelope and various other Artiodactyla as vertebrate hosts. Infection with bluetongue virus (BTV) is inapparent in the vast majority of animals but can cause fatal disease in a proportion of infected sheep, deer and wild ruminants. Infection of cattle with BTV does not usually result in clinical signs, with the exception of BTV8 infection in Europe. Cattle are particularly significant in the epidemiology of the disease due to the prolonged viraemia in the absence of clinical disease. Clinical signs of BT are mainly attributable to vascular permeability and include fever, hyperaemia and congestion, facial oedema and haemorrhages, and erosion of the mucous membranes. However in mild cases of the disease, a transitory hyperaemia and slight ocular and nasal discharge may be observed.

Identification of the agent: *BTV is a member of the Orbivirus genus of the family Reoviridae, one of the 22 recognised species or serogroups in the genus. The BTV species, or serogroup, contains 26 recognised serotypes. The serotype of individual viruses in each species is identified on the basis of neutralisation tests and different strains within a serotype are identified by sequence analysis. Complete BTV particles consist of a double-shelled icosahedron containing double-stranded RNA. The outer layer includes two proteins, one of which, VP2, is the major determinant of serotype specificity. The inner shell and core contains two major and three minor proteins and ten double-stranded RNA genetic segments. VP7 located in the inner shell is the major core protein possessing the species or serogroup-specific antigens. Virus identification traditionally requires isolation and amplification of the virus in embryonated chicken eggs, Culicoides cells, tissue culture or inoculations of susceptible ruminants and the subsequent application of serogroup- and serotype-specific tests. Reverse-transcription polymerase chain reaction (RT-PCR) technology has permitted rapid amplification of BTV cDNA in clinical samples, and RT-PCR-based procedures are now available. Real-time PCR techniques allow for more rapid and sensitive testing, and methods have been validated and published. These procedures augment the classical virological techniques to provide information on virus serogroup, serotype and toptotype.*

Serological tests: *Serological responses appear some 7–14 days after BTV infection and are generally long-lasting. A monoclonal antibody-based competitive enzyme-linked immunosorbent assay to specifically detect anti-BTV (serogroup) antibodies is recommended. Procedures to identify and quantify the BTV serotype antibodies are more complex, being typically based on neutralisation tests.*

Requirements for vaccines: *Vaccination is used in several countries to limit direct losses, minimise the circulation of BTV and allow safe movement of animals. Live attenuated vaccines are inexpensive to produce in large quantities, generate protective immunity after a single inoculation and have proven effective in preventing clinical BT disease. Adverse consequences are depressed milk production in lactating sheep, and abortion/embryonic death and teratogenesis in offspring from pregnant females that are vaccinated during the first half of gestation. Another risk associated with the use of live attenuated vaccines is their potential for spread by vectors, with eventual reversion to virulence or reassortment of vaccine virus genes with those of wild-type virus strains. The frequency and significance of these events remain poorly defined, but transmission of vaccine strains by vector Culicoides in the field has already been documented in Europe.*

A. INTRODUCTION

Bluetongue (BT) is an infectious, non-contagious, vector-borne viral disease that affects wild and domestic ruminants. Midges of just a few species in the genus *Culicoides* (the insect host) (Standfast *et al.*, 1985) transmit bluetongue virus (BTV) among susceptible ruminants, having become infected by feeding on viraemic animals (the vertebrate host). In many parts of the world infection has a seasonal occurrence (Verwoerd & Erasmus, 2004). BTV does not establish persistent infections in ruminants, and survival of BTV in the environment is associated with insect factors (Lunt *et al.*, 2006; MacLachlan, 2004). Epidemiological systems (episystems) delimited by vector species and their natural history (Gibbs & Greiner, 1994) are considered to determine the global distribution of BTV). Recent observations in Europe and the USA indicate that strains of BTV can move between episystems and adapt to different species of vector midges.

The vertebrate hosts for BTV include both domestic and wild ruminants, such as sheep, goats, cattle, buffaloes, deer, most species of African antelope and other *Artiodactyla* such as camels. Although antibodies to BTV and virus antigen or nucleic acid or live virus has been demonstrated in some carnivores, felids, black and white rhinoceroses and elephants, the role of non-ruminant species in BTV epidemiology is considered minimal. The outcome of infection ranges from unapparent in the vast majority of infected animals, especially wild African ruminants, cattle and goats, to serious or fatal in a proportion of infected sheep, goats, deer and some wild ruminants (Verwoerd & Erasmus, 2004). However a higher incidence of clinical disease has been observed in cattle infected with BTV 8 in Europe. Some breeds of sheep are more susceptible to disease than others, with the result that in some countries BTV infections of livestock can occur unobserved and be detected only by active surveillance (Daniels *et al.*, 2004).

Clinical signs of disease in sheep vary markedly in severity, influenced by the type or strain of the infecting virus, husbandry factors as well as by breed (Verwoerd & Erasmus, 2004). In severe cases there is an acute febrile response characterised by hyperaemia and congestion, leading to oedema of the face, eyelids and ears, and haemorrhages and erosions of the mucous membranes. The tongue may show intense hyperaemia and become oedematous, protrude from the mouth and, in severe cases become cyanotic. Hyperaemia may extend to other parts of the body particularly the coronary band of the hoof, the groin, axilla and perineum. There is often severe muscle degeneration. Breaks in the wool may occur associated with pathology in the follicles. A reluctance to move is common and torticollis may occur in severe cases. In fatal cases the lungs may show interalveolar hyperaemia, severe alveolar oedema and the bronchial tree may be filled with froth. The thoracic cavity and pericardial sac may contain varying quantities of plasma-like fluid. Most cases show a distinctive haemorrhage near the base of the pulmonary artery (Verwoerd & Erasmus, 2004).

Control of BTV in animals is covered in Chapter 8.3 of the OIE *Terrestrial Animal Health Code*. Virus may be introduced to a free area via infected insects, live ruminants or in contaminated products that are then transmitted to susceptible ruminants. If appropriate *Culicoides* spp. competent as vectors are present, virus can then be transmitted to other hosts. BTV is not known to cause disease in humans under any conditions.

Taxonomically, BTV is classified as a species or serogroup in the *Orbivirus* genus in the family *Reoviridae*, one of 22 recognised species in the genus that also includes epizootic haemorrhagic disease virus (EHDV), equine encephalosis and African horse sickness (AHS) viruses (Attoui *et al.*, 2012). There is significant immunological cross-reactivity among members of the BTV serogroup (Monaco *et al.*, 2006). Within species, individual members are differentiated on the basis of genotype and neutralisation tests, and currently 26 serotypes of BTV are recognised including Toggenburg virus (BTV 25) and a serotype 26 from Kuwait.

BTV particles are composed of three protein layers. The outer capsid layer contains two proteins, VP2 and VP5. VP2 is the major neutralising antigen and determinant of serotype specificity. Removal of the outer VP2/VP5 layer leaves a bi-layered icosahedral core particle that comprises an outer layer composed entirely of capsomeres of VP7 and a complete inner capsid shell (the subcore layer), which surrounds the 10 dsRNA genome segments and minor structural proteins. VP7 is a major determinant of serogroup specificity and the site of epitopes used in competitive enzyme-linked immunosorbent assay (C-ELISA) to detect anti-BTV antibodies (Mertens *et al.*, 2005). VP7 can also mediate attachment of BTV to insect cells (Attoui *et al.*, 2012).

Genetic sequencing of BTVs allows for additional differentiation and analysis of strains apart from serotyping (Gould, 1987; McColl & Gould, 1991; Pritchard *et al.*, 1995; Wilson *et al.*, 2000). Even for strains within the one serotype it is possible to identify the likely geographical origin (topotype) (Gould, 1987; Potgieter *et al.*, 2005). Identification of apparent associations between some genotypes of virus and some vector species has led to further development of the concept of viral-vector ecosystems (Daniels *et al.*, 2004; Gibbs & Greiner, 1994; MacLachlan, 2004). Recent movements of several BTV serotypes between vector species and into new geographical regions indicate that a more complete understanding of BTV epidemiology is required.

There is no known risk of human infection with BTV. Biocontainment measures should be determined by risk analysis as described in Chapter 1.1.4 *Biosafety and biosecurity: Standard for managing biological risk in the veterinary laboratory and animal facilities*.

B. DIAGNOSTIC TECHNIQUES

Table 1. Test methods available for the diagnosis of bluetongue and their purpose

Method	Purpose					
	Population freedom from infection	Individual animal freedom from infection prior to movement	Contribute to eradication policies	Confirmation of clinical cases	Prevalence of infection – surveillance	Immune status in individual animals or populations post-vaccination
Agent identification¹						
Real-time RT-PCR	–	+++	–	+++	++	–
RT-PCR	–	+++	–	+++	++	–
Classical virus isolation	–	+++	–	+++	–	–
Detection of immune response						
C-ELISA (serogroup specific)	++	+++	++	–	++	++
VN (serotype specific)	++	+++	++	–	++	++
AGID	+	–	+	–	+	+
CFT	+	–	+	–	+	+

Key: +++ = recommended method; ++ = suitable method; + = may be used in some situations, but cost, reliability, or other factors severely limits its application; – = not appropriate for this purpose.

Although not all of the tests listed as category +++ or ++ have undergone formal validation, their routine nature and the fact that they have been used widely without dubious results, makes them acceptable.

RT-PCR = reverse-transcription polymerase chain reaction; C-ELISA = competitive enzyme-linked immunosorbent assay; VN = virus neutralisation; AGID = agar gel immunodiffusion; CFT = complement fixation test.

1. Identification of the BTV agent

1.1. *In-vitro* and *in-vivo* cultures

The same diagnostic procedures are used for domestic and wild ruminants. A number of virus isolation systems for BTV are in common use, but the most sensitive method is the inoculation of embryonated chicken eggs (ECE). Primary inoculation of cell cultures such as the KC cell line (a cell-line derived from *C. variipennis* midges), has been proven to be very sensitive (McHolland & Mecham, 2003). Inoculation of sheep may also be a useful approach if the titre of virus in the sample is very low, as may be the case several weeks after virus infection, or where laboratory facilities are not available. Attempts to isolate virus in cultured cells *in vitro* may be more convenient, but the success rate is frequently much lower than that achieved with *in-vivo* systems (Gard *et al.*, 1988). Specimens for virus isolation include unclotted blood from suspected viraemic animals, blood clots after separation of serum, spleen or lymph nodes collected at necropsy of clinical cases, or midges.

¹ A combination of agent identification methods applied on the same clinical sample is recommended.

1.1.1. Isolation in embryonated chicken eggs

- i) Blood is collected from suspected viraemic animals into an anticoagulant such as EDTA (ethylamine diamine tetra-acetic acid), heparin or sodium citrate, and the blood cells are washed three times with sterile phosphate buffered saline (PBS). Washed cells are re-suspended in PBS or isotonic sodium chloride and either stored at 4°C or used immediately for attempted virus isolation. Tissue and midge suspension can be also prepared and stored as described above or immediately used.
- ii) For long-term storage where refrigeration is not possible blood samples are collected in oxalate–phenol–glycerin. If samples can be frozen, they should be collected in buffered lactose peptone or 10% dimethyl sulphoxide and stored at –70°C or colder. The virus is not stable at –20°C. BTV has remained viable for several months in whole blood in anticoagulant stored at 4°C.
- iii) In fatal cases, spleen and lymph nodes are the preferred organs for virus isolation attempts. Organs and tissues should be kept and transported at 4°C to a laboratory where they are homogenised in PBS or isotonic saline (1/10), centrifuged at 1500 rpm for 10 minutes, and filtered (0.2–0.4 µm). The tissue suspensions can be used as described below for blood cells.
- iv) Washed blood cells are re-suspended in distilled water or sonicated in PBS and 0.1 ml amounts inoculated intravascularly into 5–12 ECE that are 9–12 days old. This procedure requires practice. Details are provided by Clavijo *et al.* (2000).
- v) The eggs are incubated in a humid chamber at 32–33.5°C and candled daily. Any embryo deaths within the first 24 hours post-inoculation are regarded as nonspecific.
- vi) Embryos that die between days 2 and 7 are retained at 4°C and embryos remaining alive at 7 days are killed. Infected embryos may have a haemorrhagic appearance. Dead embryos and those that live to 7 days are homogenised as two separate pools. Whole embryos, after removal of their heads, or pooled organs such as the liver, heart, spleen, lungs and kidney, are homogenised and the debris removed by centrifugation.
- vii) Virus in the supernatant may be identified either directly as described in Section 1.2 below or after further amplification in cell culture, as described in Section 1.1.2.

1.1.2. Isolation in cell culture

Virus isolation may be attempted in BTV susceptible cell cultures such as mouse L, baby hamster kidney (BHK-21), African green monkey kidney (Vero) or *Aedes albopictus* clone C6/36 (AA). The efficiency of isolation is often significantly lower following inoculation of cultured cells with diagnostic samples compared with that achieved in ECE. Highest recovery rates are achieved by primary isolation of virus in ECE followed by passage in AA cells or mammal cells for further replication of virus. Successful virus isolation has also been reported using primary isolation in cells derived from *Culicoides sonorensis* free of BTV and *Culicoides* viruses and designated as KC or CuVa cells (McHolland & Mecham, 2003; Wechsler *et al.*, 1989). In case of passage in AA, KC or CuVa cells, additional passages in mammalian cell lines such as BHK-21 or Vero are usually performed. A cytopathic effect (CPE) is not necessarily observed in AA, KC or CuVa cells but appears in mammalian cells. Cell monolayers are monitored for the appearance of CPE for 5 days at 37°C in 5% CO₂ with humidity. If no CPE appears, a second passage is made in the mammalian cell culture. Isolated BTV may be detected after each ECE or cell culture passage by antigen detection or polymerase chain reaction (PCR) techniques.

1.1.3. Isolation in sheep

This procedure for isolation of BTV is not routinely used, but is useful where laboratory facilities are not available or where there is a requirement to propagate virus without using *in-vitro* isolation systems.

- i) Sheep are inoculated with washed cells from 10 ml to 500 ml of blood, or 10–50 ml tissue suspension. Inocula are administered subcutaneously in 10–20 ml aliquots. Large volumes may aid in the virus isolation attempts and should be administered intravenously.
- ii) The sheep are held for 28 days and checked daily for pyrexia and weekly for antibody response using serological tests such as the C-ELISA as described below. Sheep blood collected at 7–14 days post-inoculation will usually contain the isolated virus, which can be stored viable at 4°C or –70°C and detected and characterised using the methods described in Sections B.1.2 and B.1.3 below.

1.2. Virus detection and characterisation

The success of virus isolation techniques is assessed by testing for the presence of BTV in the cell culture supernatants, embryo tissues or inoculated animal's blood using any of a number of detection systems. Prior to the advent of PCR techniques immunodetection techniques were used. Currently testing of the isolation mediums by real-time PCR is the preferred screening method. Hence virus in the supernatant may be identified either directly by C-ELISA, reverse-transcription PCR (RT-PCR) or real-time RT-PCR, as described in Section B.1.3 below.

Detection and characterisation is typically a step-wise process, with serogroup-specific tests used initially to detect the presence of a BTV. Subsequent genotype and serotype identification of BTV isolates provides valuable epidemiological information and is critical for the implementation of vaccines or for vaccine development. RT-PCR assays employing serotype-specific primers will provide the most rapid and specific information regarding isolate serotype (Johnson *et al.*, 2000; Maan *et al.*, 2012; Mertens *et al.*, 2007).

Genotyping for molecular epidemiology can be based on RT-PCR tests and sequencing of the amplicon. Different laboratories have standardised on several different gene sequences for this purpose. Where available, full genome sequencing may also be performed to provide serotype, as well as other unique sequence information of isolates.

Neutralisation procedures using individual serotype antisera may also be employed for serotyping, although some serotypes are cross-reactive and interpretation can be difficult. For laboratories without serotyping capabilities, BTV isolates may be submitted to any of several OIE BT Reference Laboratories for serotyping of isolates.

1.2.2. Immunological serogrouping of viruses

Orbivirus isolates are typically serogrouped on the basis of their reactivity with specific standard antisera that detect proteins, such as VP7, that are conserved within each serogroup. The cross-reactivity between members of the BT and EHD serogroups raises the possibility that an isolate of EHDV could be mistaken for BTV on the basis of a weak immunofluorescence reaction with a polyclonal anti-BTV antiserum. Polyclonal and monoclonal antibodies used for serogrouping BTV isolates must be characterised as appropriate for the purpose. There exists significant VP7 variation within BTV, as well as antigenic relatedness between other closely related orbiviruses, such as EHDV, that will influence antibodies binding in different assay formats (IFA, ELISA, AGID). For this reason, a BT serogroup-specific MAb can be used. A number of laboratories have generated such serogroup-specific reagents. Commonly used methods for the identification of viruses to serogroup level are as follows.

i) *Immunofluorescence*

Monolayers of BHK or Vero cells on chamber slides (glass cover-slips) are infected with either tissue culture-adapted virus or virus in ECE lysates. After 24–48 hours at 37°C, or after the appearance of a mild CPE, infected cells are fixed with agents such as paraformaldehyde, acetone or methanol, dried and viral antigen detected using anti-BTV antiserum or BTV-specific MAbs and standard immunofluorescent procedures.

ii) *Antigen capture enzyme-linked immunosorbent assay*

Viral antigen in ECE and culture medium harvests, infected insects and sheep blood may be detected directly. In this technique, virus derived proteins are captured by antibody adsorbed to an ELISA plate and bound materials detected using a second antibody. The capture antibody may be polyclonal or a serogroup-specific MAb. Serogroup-specific MAbs and polyclonal antibody raised to baculovirus-expressed core particles have been used successfully to detect captured virus.

1.2.2. Serotyping by virus neutralisation

Neutralisation tests are type specific for the currently recognised BTV serotypes that have been isolated in culture and can be used to serotype a virus isolate or can be modified to determine the specificity of antibody in sera. In the case of an untyped isolate, the characteristic regional localisation of BTV serotypes can generally obviate the need to attempt neutralisation by antisera to all isolated serotypes, particularly when endemic serotypes are well known.

There is a variety of tissue culture-based methods available to detect the presence of neutralising anti-BTV antibodies. Cell lines commonly used are BHK, Vero and L929. Three methods to serotype BTV are outlined briefly below. There is also a fluorescence inhibition test, not described. BTV serotype-specific antisera generated in guinea-pigs or rabbits have been reported to have less serotype cross-reactivity than those made in cattle or sheep. It is important that antiserum controls be included.

i) *Plaque reduction*

The virus to be serotyped is diluted to contain approximately 100 plaque-forming units (PFU), and incubated with either no antiserum (virus control) or with serial dilutions of individual standard antisera to a panel of BTV serotypes. Virus/antiserum mixtures are added to monolayers of cells. After adsorption and removal of inoculum, monolayers are overlaid with agarose. The neutralising antibody titres are determined as the reciprocal of the serum dilution that causes a fixed reduction (e.g. 90%) in the number of PFU. The unidentified virus is considered serologically identical to a standard serotype if the latter is run in parallel with the untyped virus in the test, and is similarly neutralised.

ii) *Plaque inhibition*

Tests are performed in 90 mm diameter Petri dishes containing confluent cell monolayers that are infected with approximately 5×10^4 PFU standard or untyped virus. After adsorption and removal of inoculum, monolayers are overlaid with agarose. Standard anti-BTV antisera are added to individual filter paper discs and placed on the agarose surface. Dishes are incubated for at least 4 days. A zone of virus neutralisation, with concomitant survival of the cell monolayer, will surround the disc containing the homologous antiserum.

iii) *Microtitre neutralisation*

Approximately 100 TCID₅₀ (50% tissue culture infective dose) of the standard or untyped virus is added in 50 µl volumes to test wells of a flat-bottomed microtitre plate and mixed with an equal volume of standard antiserum serially diluted in tissue culture medium. Approximately 10^4 cells are added per well in a volume of 100 µl, and after incubation for 4–6 days, the test is read using an inverted microscope. Wells are scored for the degree of CPE observed. Those wells that contain cells only or cells and antiserum, should show no CPE. In contrast, wells containing cells and virus should show 75–100% CPE. The unidentified virus is considered to be serologically identical to a standard BTV serotype if both are neutralised in the test to a similar extent.

1.3. Molecular methods – detection of nucleic acid

RT-PCR techniques provide rapid identification of BT viral nucleic acid in blood and other tissues of infected animals. Importantly, RT-PCR-based diagnostics should be interpreted with caution because the RT-PCR procedure will detect virus-specific nucleic acid after the virus is no longer viable and capable of establishing a new infection in either insects or mammalian hosts. Hence a positive RT PCR result, does not necessarily indicate the presence of infectious virus (MacLachlan *et al.*, 1994).

Multiple RT-PCR formats are available that can be used to detect BTV specifically to 'serogroup' Orbiviruses and to 'serotype' BTV. These molecular approaches are much more rapid than traditional virological and immunological approaches, which may require up to 4 weeks to generate information on serogroup and serotype.

The nucleic acid sequence of cognate BTV genes may differ with the geographical area of virus isolation (Gould, 1987). This has provided a unique opportunity to complement studies of BTV epidemiology by providing information on the potential geographical origin of virus isolates, a process termed genotyping or topotyping. While it appears likely that sequencing of BTV isolates from different parts of the world may permit finer discrimination of geographical origin, the relationship between sequence and geographical origin may not be straightforward. This sequencing information is important and all data regarding BTV segment sequences should be made widely available by submitting the data to officially recognised web sites (e.g. <http://oiebtnet.izs.it>).

The web sites provide phylogenetic tree analyses of BTV isolates based on the sequence of RNA segments. These compiled data will provide a resource for epidemiological studies, the identification of new isolates and the design of new primers for further RT-PCR and possibly serotype-specific assays for BTV.

The capacity of RT-PCR assays to detect very small numbers of nucleic acid molecules means that such tests are exquisitely sensitive to contamination by extraneous nucleic acids. The latter may include any primers in use in the laboratory or previously amplified polynucleotides. It is critical therefore to have a 'clean' area containing all equipment necessary for reagent and test preparation and a separate area with its own equipment for amplification. Impervious gloves should be worn and changed frequently at all stages of the procedure, particularly after working with sample RNA or amplified DNA. This will help protect reagents and samples from contamination by ubiquitous RNases and other agents and from cross-contamination by DNA. The possibility of false positives, due to sample contamination, highlights the importance of sequencing RT-PCR products to determine, for example, if the amplified sequence is identical to or different from that of the positive control. False negatives, due for example to poor sample quality or inappropriate primers, may be identified following the failure to amplify both BTV and a host gene, such as globin, from extracts of infected cells. This is covered in more detail in Chapter 1.1.6 *Principles and methods of validation of diagnostic assays for infectious diseases*.

There are many RT-PCR assays currently in use that use different extraction methods, reverse transcriptases, amplification enzymes, primers and conditions. Technology is changing rapidly. Also the genetic diversity of the BTV genes makes the choice and validation of RT-PCR assays conditional on its application in a regional setting. This is also the case for real-time PCR, which will be developed for use in routine molecular diagnosis of BTV in the future. Therefore the procedures listed below are examples only. Increasingly it will be more important to maintain diagnostic testing under accreditation to an international standard such as ISO/IEC 17025 and participate in proficiency testing. Systems for offering proficiency testing for RT-PCR tests are being developed in a number of countries.

Two BTV RT-PCR assays are presented here: a real-time assay (Hofmann *et al.*, 2008), targeting the NS3 genome; and a conventional nested assay targeting the NS1 genome, using primers designed by Katz *et al.* (1993). The nested assay has been successfully used for over 20 years and is capable of detecting serotypes 1–24 and 26 (serotypes 25 not tested) from multiple species. The nested assay may be beneficial for laboratories without the capacity to perform real-time PCR. The real-time RT-PCR assay presented below has been tested at several laboratories world-wide and has been found capable of detecting all serotypes of BTV, and equals or surpasses the sensitivity of the nested assay while providing rapid, quantitative detection of BTV without the contamination risks associated with nested PCR assays.

1.3.1. Real-time reverse-transcription polymerase chain reaction

Real-time RT-PCR methods provide sensitive and rapid detection of BTV in a one-step procedure. Advantages of real-time methods over traditional PCR methods include rapidity of testing, quantitation of the virus present, and the reduced opportunity for contamination to occur as no post-amplification handling such as gel electrophoresis is needed. The real-time RT-PCR assays are the tests of choice for diagnosis.

The method presented here is an adaption from Hofmann *et al.* (2008) and is capable of detecting all known BTV serotypes and strains currently circulating. The assay targets BTV segment 10 (NS3). The procedure given may require modification to accommodate individual laboratory or different RT-PCR kit requirements.

i) RNA extraction from blood, tissue samples, and midges

Commercial kits are widely available; the RNA extraction step can be performed according to the procedures specified in each kit.

ii) Real-time reverse-transcription polymerase chain reaction

Kits for the one-step real-time PCR are available commercially. Below are some basic steps as described by Hofmann *et al.* (2008), which can be modified depending upon local/case-specific requirements, kits used and equipment available.

Primer and probe sequences for the detection of BTV species viruses:

BTV_IVI_F 5'-TGG-AYA-AAG-CRA-TGT-CAA-A-3'

BTV_IVI_R 5'-ACR-TCA-TCA-CGA-AAC-GCT-TC-3'

BTV_IVI_P 5'FAM-ARG-CTG-CAT-TCG-CAT-CGT-ACG- C-3' BHQ1

a) Primer stock solutions are diluted to a working concentration of 20 pmol/μl, whereas probe is diluted to a working concentration of 5 pmol/μl.

- b) A test plate layout should be designed and loaded into the real-time PCR machine software. Using the layout as a guide, 0.5 µl of each primer working stock (20 pmol/µl) is added to each well that will go on to contain RNA samples, positive or negative controls. The plate is held on ice.

Note: PCR plates can be replaced with tubes or strips as appropriate.

- c) 2 µl of RNA samples, including test and positive and negative controls, are added to appropriate wells of the plate following the layout (note: these wells already contain primers from step b).
- d) Heat denaturation: 95°C for 5 minutes, hold on ice for further 3 minutes.
- e) An appropriate volume of real-time one-step RT-PCR master mix for the number of samples to be tested is prepared, following the manufacturer's instructions. Probe should be included in the master mix to give a final concentration of 0.2 pmol/µl per sample.
- f) 22 µl of master mix is distributed in each well on the PCR plate containing the denatured primers and RNA.
- h) The plate is placed in a real-time thermal cycler programmed for reverse transcription and cDNA amplification/fluorescence detection as suggested by the manufacturers. The following thermal profile is an example:

	48°C	x30 minutes
	95°C	x2 minutes
50 cycles:	95°C	x15 seconds
	56°C	x30 seconds
	72°C	x30 seconds

1.3.2. Reverse-transcription polymerase chain reaction – nested PCR

Select RT-PCR (reverse-transcription and first stage amplification) and PCR (nested amplification) kits of choice to perform the nested assay. The assay presented below for illustration uses the parameters associated with a specific kit. Adjust the PCR parameters according to the manufacturer's recommendations for the specific kits to be used.

The nested assay employs the use of the following primers:

First stage amplification (outer) (dilute to 25 pmol/µl; final concentration in PCR is 0.6 µM each):

FW: GTT-CTC-TAG-TTG-GCA-ACC-ACC
RV: AGG-CCA-GAC-TGT TTC-CCG-AT

Nested amplification (dilute to 25 pmol/µl; final concentration in PCR is 0.5 µM each):

nFW: GCA-GCA-TTT-TGA-GAG-AGC-GA
nRV: CCC-GAT-CAT-ACA-TTG-CTT-CCT

- i) Prepare the first stage amplification mixture (one-step RT-PCR kit) of the following reagents (per sample):

Nuclease-free water	11.8 µl
5x one-step RT-PCR buffer	5.0 µl
dNTP mix	1.0 µl
Enzyme	1.0 µl
FW primer (25 pmol/µl)	0.6 µl
RV primer (25 pmol/µl)	0.6 µl

- ii) Dispense 20 µl of the mixture into each PCR tube included in the assay. Add 5 µl of sample or control denatured RNA (described above) to the appropriate tube. Place tubes in a thermal cycler and run the following programme:

Reverse transcription	50°C	30 minutes
Taq activation	95°C	15 minutes

Followed by 35 cycles of:

Denature:	94°C	45 seconds
Anneal:	58°C	45 seconds
Extension	72°C	60 seconds (final extension 10 minutes)

- iii) Prepare a nested PCR mixture (HotStarTaq DNA Polymerase kit) of the following reagents (per sample):

Nuclease-free water	40.75 µl
10xHotStar buffer	5.0 µl
dNTP mix	1.0 µl
HotStarTaq	0.25 µl
nFW primer (25 pmol/µl)	1.0 µl
nRV primer (25 pmol/µl)	1.0 µl

- iv) Dispense 49 µl of the mixture into each PCR tube. Transfer 1.0 µl of the amplified DNA from the first stage reaction (step 2) to the appropriate nested tube. Change gloves between samples, and use caution when transferring the DNA to avoid cross contamination of samples. Place tubes in a thermal cycler and run the following programme:

Taq activation	95°C	15 minutes
Followed by 28 cycles of:		
Denature:	94°C	45 seconds
Anneal:	62°C	45 seconds
Extension:	72°C	1 minute (final extension 10 minutes)

Perform gel electrophoresis followed by ultraviolet visualisation on the nested PCR product. The positive control(s) and any positive samples will have a 101 base pair band. Negative controls and negative samples should not have a visible band. Positive samples may be sequenced for verification.

2. Serological tests

Anti-BTV antibodies generated in infected animals can be detected in a variety of ways that vary in sensitivity and specificity. Both serogroup-specific and serotype-specific antibodies are elicited and if the animal was not previously exposed to BTV, the neutralising antibodies generated are specific for the serotype of the infecting virus. Multiple infections with different BTV serotypes lead to the production of antibodies capable of neutralising serotypes to which the animal has not been exposed.

2.1. Complement fixation

A complement fixation test (CFT) to detect BTV antibodies was widely used until 1982, when it was largely replaced by the AGID test although the CFT is still used in some countries.

2.2. Agar gel immunodiffusion

It must be recognised that a major disadvantage of the AGID used for BT is its lack of specificity in that it does not exclusively differentiate between antibodies to the BT and EHD serogroups hence it cannot be used definitively to detect antibodies to BTV as a positive reaction may have been the result of an infection to another *Orbivirus* species. However the AGID test is simple to perform and the antigen used in the assay relatively easy to produce. Since 1982, the test has been one of the standard testing procedures for international movement of ruminants, but is no longer considered sufficiently accurate for use in the support of international trade. AGID positive sera should be retested using a BT serogroup-specific assay if BTV specificity is required. A preferred test, a C-ELISA is described in Section B.2.3.

2.3. Competitive enzyme-linked immunosorbent assay

The BT competitive or blocking ELISA was developed to measure BTV-specific antibody without detecting cross-reacting antibody to other Orbiviruses (Afshar *et al.*, 1989; Lunt *et al.*, 1988). The specificity is the result of using one of a number of BT serogroup-reactive MABs. These antibodies were derived in a number of laboratories, and although different, all appear to bind to the amino-terminal region of the major core protein VP7. In the C-ELISA, antibodies in test sera compete with the MABs for binding to antigen. The following procedure for the C-ELISA has been standardised after comparative studies in a number of international laboratories.

2.3.1. Test procedure

There are several test procedures described; this is an example of one BT ELISA procedure.

- i) First, 96-well microtitre plates are coated at 4°C overnight or at 37°C for 1 hour with 50–100 µl of either tissue culture-derived sonicated cell culture antigen or the major core antigen VP7 expressed in either *Baculovirus* (Oldfield *et al.*, 1990) or yeast (Martyn *et al.*, 1990) and diluted in 0.05 M carbonate buffer, pH 9.6.
- ii) The plates are washed five times with PBST (0.01 M PBS containing 0.05% or 0.1% Tween 20, pH 7.2).
- iii) Next, 50 µl of test sera is added in duplicate at a single dilution, either 1/5 (Afshar *et al.*, 1989) or 1/10 (Lunt *et al.*, 1988) in PBST containing 3% bovine serum albumin (BSA).
- iv) Immediately, 50 µl of a predetermined dilution of MAb diluted in PBST containing 3% BSA is added to each well. MAb control wells contain diluent buffer in place of test serum.
- v) Plates are incubated for 1 hour at 37°C or 3 hours at 25°C, with continuous shaking.
- vi) After washing as described above, wells are filled with 100 µl of an appropriate dilution of horseradish peroxidase-labelled rabbit anti-mouse IgG (H+L) in PBST containing 2% normal bovine serum.
- vii) Following incubation for 1 hour at 37°C, the conjugate solution is discarded and plates are washed five times using PBS or PBST. Wells are filled with 100 µl substrate solution containing 1.0 mM ABTS (2,2'-azino-bis-[3-ethylbenzothiazoline-6-sulphonic acid]), 4 mM H₂O₂ in 50 mM sodium citrate, pH 4.0, and the plates are shaken at 25°C for 30 minutes. (Other substrates may be used and the reaction continued with shaking for an appropriate length of time to permit colour development.)
- viii) The reaction is stopped by addition of a stopping reagent, such as sodium azide.
- ix) After blanking the ELISA reader on wells containing substrate and stopping reagent, the absorbance values are measured at 414 nm. Results are expressed as per cent inhibition and are derived from the mean absorbance values for each sample by the following formula:
$$\% \text{ inhibition} = 100 - [(\text{mean absorbance test sample})/(\text{mean absorbance MAb control}) \times 100].$$

NB: Some laboratories prefer to use a negative control serum that has previously been shown to have a percentage inhibition of zero as an alternative to the MAb control.
- x) Percentage inhibition values >50% are considered to be positive. Inhibition between 40% and 50% is considered to be suspicious. The results of the test sera duplicates can vary as long as the results do not lie either side of the positive cut-off.
- xi) Strong and weak positive sera and a negative serum should be included on each plate. The weak positive should give 60–80% inhibition and the negative should give less than 40% inhibition.

A number of commercially produced C-ELISAs based on recombinant VP-7 and anti-VP-7 MAb are now available. These commercial assays are routinely used in many laboratories across the world and have been proved to be fit for purpose in ring-trials (Batten *et al.*, 2008). Formal acceptance for trade purposes should depend on adoption of individual kits to the OIE Register.

Genetic divergence of certain BTV strains (e.g. different regional groups or topotypes) may affect the nature of serogroup-reactive antibodies. It is therefore possible that diagnostic characteristics for antibody detection are not uniform for all viruses encompassed by the serogroup. This should be addressed in considerations of fitness for purpose.

2.4. Indirect ELISA

An indirect ELISA for bulk milk samples has been shown to be reliable and useful for surveillance purposes (Kramps *et al.*, 2008). It should be validated for relevant serotypes before use.

2.5. Virus neutralisation serology

VN serology is able to identify serotype-specific neutralising antibodies as well as determine their titre. It is an important additional test in endemic areas where multiple serotypes are likely to be present. Its capability to identify the serotype involved in an outbreak is essential for putting in place appropriate control measures such as vaccination. It is also useful for epidemiological surveillance, transmission studies and for determining useful antibody response to vaccination. Cross-neutralising antibodies can develop in animals that have experienced BTV infection. In particular, infection with a second serotype

can broaden the neutralising antibody response to include antibodies to serotypes to which the animal has not been exposed. The application of VN serology is frequently most useful in conjunction with other virological investigations that, in combination, can provide a more definitive basis for resolving serotype distribution.

2.5.1. Test procedure

Several methods to determine titre and serotype of BTV have been described; here the procedure that has been standardised after comparative studies in a number of international laboratories is briefly outlined. Cell lines commonly used are BHK and Vero. BTV serotype-specific antisera generated in guinea-pigs or rabbits have been reported to have less serotype cross-reactivity than those made in cattle or sheep. It is important that antiserum controls be included.

- i) 50µl of serial sera dilutions, from 1/10 to 1/1280, are added to each test well of flat-bottomed microtitre plates and each well is mixed with an equal volume of OIE standard reference BTV serotypes (100–300 TCID₅₀). The plates are incubated at 37°C in 5% CO₂.
- ii) After 1 hour of incubation, approximately 10⁴ Vero or BHK-21 cells are added per well in a volume of 100 µl, of MEM (minimal essential medium) containing antibiotics and, after incubation for 3–5 days, the test is read using an inverted microscope.
- iii) Wells are scored for the degree of CPE observed. A sample is considered positive when it shows more than 75% of CPE neutralisation at the lowest dilution (1/10). The serum titre represents the highest serum dilution capable of neutralising more than 75% CPE in the tissue culture.

C. REQUIREMENTS FOR VACCINES

1. Background

Both live attenuated and inactivated BTV vaccines are currently available for use in sheep and sheep and cattle, respectively. Recombinant BT vaccines based on various approaches are under development but none has been licensed and these vaccines will not be addressed here. In South Africa live attenuated vaccines have been used for over 50 years and are known to induce an effective and lasting immunity (Erasmus, 1975). Live attenuated vaccines are produced by adapting BTV field isolates to growth *in vitro* through serial passages in tissue culture or in ECEs. Stimulation of a strong antibody response by these vaccines is directly correlated to their ability to replicate in the vaccinated host. Live attenuated vaccines are cheap to produce in large quantities; they generate protective immunity after a single inoculation and have proven effective in preventing clinical BT disease in the areas where they are used (Dungu *et al.*, 2004). However, live attenuated BTV vaccines suffer from a variety of documented or potential drawbacks, including under-attenuation, the impact of which may vary with different breeds of sheep. The frequency and significance of these events remain poorly defined but transmission of vaccine strains by vector midges has already been documented in the USA, South Africa and Europe (Ferrari *et al.*, 2005; MacLachlan *et al.*, 1985; Monaco *et al.*, 2006; Venter *et al.*, 2004).

Guidelines for the production of veterinary vaccines are given in Chapter 1.1.8 *Principles of veterinary vaccine production*. The guidelines given below and in chapter 1.1.8 are intended to be general in nature and may be supplemented by national and regional requirements.

2. Outline of production and minimum requirements for vaccines

2.1. Characteristics of the seed

See chapter 1.1.8 for general requirements for master seeds and allowable passages for vaccine production.

2.1.1. Biological characteristics of the master seed

For live attenuated vaccines, the master or primary virus seed is prepared from a single plaque of serially passaged, attenuated BTV. Vaccine viruses have been attenuated by either passage in ECE, tissue culture cells or a combination of both. Each primary seed virus lot should also be tested for transmissibility and reversion to virulence prior to vaccine manufacture. Samples of vaccine prepared from secondary seed virus at the maximum permitted passage level should be tested in sheep for avirulence, safety and immunogenicity.

For inactivated vaccines, the issues of attenuation do not apply, and the approach adopted has been to use field strains of low passage level with the intent of achieving high antigenicity.

2.1.2. Quality criteria (sterility, purity, freedom from extraneous agents)

Primary seed virus must be free of contaminating bacteria, viruses, prions, fungi and mycoplasmas, particularly pestivirus contamination. For the latter, particular attention should be paid to the fetal bovine serum used in cell cultures, as it may be contaminated. Seed viruses must be shown to have the desired serotype specificity. BTV seed lot viruses should be sequenced and the data made available to relevant databases (Osburn, 2004). Secondary seed lots, which are used as inocula for vaccine production, are usually not more than three passages beyond the primary seed lot.

2.1.3. Validation as a vaccine strain

Live attenuated BT vaccines must be safe and efficacious, and a brief description of appropriate tests for these parameters is given below. In addition, attenuated viruses should not revert to virulence during replication in vaccinated animals or be able to be transmitted from such animals by insect vectors. The latter criterion is very important because insect-mediated transmission of attenuated virus from vaccinated to non-immune animals, with the subsequent replicative steps in each host species, increases the possibility of reversion to virulence. Although tests for reversion to virulence and transmissibility are rarely, if ever performed, a brief description of what may be necessary is outlined.

There is a variation in BT susceptibility between breeds of sheep; it is important that sheep that have been proven to be susceptible to infection with BTV be used for vaccine validation.

2.1.4. Emergency procedure for provisional acceptance of new master seed virus (MSV) in the case of an epizootic

In a number of different circumstances (see Chapter 1.1.10 *Vaccine banks*) and in the event that a new or different serotype or variant of BTV results in an emergency epizootic situation that cannot be controlled by currently available vaccines, and where there is not enough time to fully test a new MSV for all extraneous agents, provisional acceptance of the new strain could be based on a risk analysis of the possibility of contamination of the antigen produced from the new MSV with extraneous agents. This risk assessment should take into account the characteristics of the process, including the nature and concentration of the inactivant for inactivated vaccines, before allowing or not the early release of the new product.

2.2. Method of manufacture

2.2.1. Procedure

Attenuation of field isolates of BTV was first achieved by serial passage in ECE. Because of the concern about transmission of the egg propagated attenuated virus, it has been recommended that animals receiving vaccines produced in ECE should not be moved internationally (Osburn, 2004). More recently, it is clear that passage in cultured cells will also result in attenuation of virulence. No studies have been done to precisely relate passage number and extent of attenuation for individual virus isolates or serotypes. To prepare attenuated virus, field isolates are adapted to cell culture and passaged *in vitro* up to 40 times or more. Ideally, a number of plaque-purified viruses are picked at this stage and each is examined to determine the level of viraemia they generate and their ability to elicit a protective immune response in vaccinated sheep. The most suitable virus is one that replicates to low titre but generates a protective immune response, and this may represent the source of vaccine primary seed stock virus.

BTV for inactivated vaccines is produced in large-scale suspension cell systems under aseptic and controlled conditions. Cell lines adapted for large scale industrial cultures are used and these are proven to be free from contaminating microorganisms. When the viral suspension virus reaches its maximum titre, cell disruption is performed and the culture is clarified and filtered. Subsequently inactivation is performed according to processes adopted by the manufacturer, such as by addition of binary ethyleneimine (BEI) or other inactivants. The process must comply with legislation relevant for the intended market, be validated to ensure complete inactivation and supported by appropriate documentation. The inactivation process should not significantly alter the immunogenic properties of the viral antigens. Purification is carried out by chromatography. The inactivated virus is then concentrated by ultrafiltration and stored. The inactivated, chromatography-purified and concentrated BTV antigens are made into vaccine by dilution in a buffer solution and addition of adjuvants.

2.2.2. Requirements for ingredients

All ingredients of animal origin, including serum and cells must be checked for the presence of viable bacteria, viruses, fungi or mycoplasma. For further details, see chapter 1.1.8 for general guidance on ingredients of animal origin. Ingredients of animal origin should be sourced from a country with negligible risk for transmissible spongiform encephalopathies (TSEs).

2.2.3. In-process controls

Virus concentration of live attenuated vaccines is assessed by infectivity and ELISAs.

For inactivated vaccines, during inactivation of the virus, timed samples are taken at regular intervals for the purpose of monitoring the rate and linearity of the inactivation process. Virus titres in the samples are determined by inoculation of BHK-21 or other appropriate cell cultures. At the end of the inactivation process, the vaccine is checked to ensure that there is no live virus.

2.2.4. Final product batch tests

i) Sterility

Every batch of vaccine should be tested for the presence of contaminant viruses, viable bacterial, fungi or mycoplasma. For example, in South Africa a pool of ten randomly selected ampoules are inoculated into soya broth and thioglycolate broth, and incubated at room temperature and 37°C for 14 days, respectively. If contaminated, the batch is disqualified.

ii) Safety

Every batch of attenuated vaccine is safety tested in newborn and adult mice, guinea-pigs and sheep. If any adverse reactions or significant signs are noted, the test is repeated. Any increase in the body temperature of the target animal that is above the level expected for the particular strain of attenuated virus under test should be regarded as symptomatic. If the results are unsatisfactory after a second attempt, the batch is disqualified.

Safety testing of inactivated vaccines is conducted in sheep to ensure side effects are not observed.

iii) Potency

Each batch is tested by inoculation of susceptible sheep. Pre-vaccination, 21- and 28-day post-vaccination sera are tested by VN assay to determine neutralising antibody levels. To be passed, the antibody titre must be equal to or higher than a set standard based on international vaccine standards.

iv) Duration of immunity

Studies with live attenuated BTV vaccine have shown that antibodies in sheep may appear before day 10 post-vaccination, reach a maximum approximately 4 weeks later and persist for well over a year. There is a temporal relationship between the increase in neutralising antibody titre and clearance of virus from the peripheral circulation. Live attenuated BTV vaccines have been in use for over 50 years and are known to induce an effective and lasting immunity in sheep (Verwoerd & Erasmus, 2004). Many serotypes of BTV may be present in endemic areas of South Africa, and polyvalent vaccines are used. The inclusion of 15 serotypes in three polyvalent vaccines that are administered sequentially sometimes means that an effective immune response is not generated to all serotypes, presumably because of the antigenic mass of individual serotype-specific antigens is small. In an attempt to broaden the response, vaccination is repeated annually (Erasmus, 1975).

Initial studies with inactivated vaccines show that antibody against BTV can be detected by day 7 post-vaccination and increase in titre to days 14–21. A second dose of vaccine boosts the titre. Data to demonstrate the expected duration of immunity are being acquired.

v) Stability

Procedures have been developed for attenuated vaccines. Stability should be tested over a period of 2 years. Vaccines in liquid and lyophilised forms are deemed to have shelf lives of 1 and 2 years, respectively. Each batch of vaccine is subjected to an accelerated shelf-

life test by storing it at 37°C for 7 days. It is then titrated and evaluated according to a set standard, as determined in the initial testing of the vaccine. Shelf life of stock aliquots stored at +4°C should be tested periodically.

2.3. Requirements for authorisation/registration/licensing

2.3.1. Manufacturing process

For registration of vaccine, all relevant details concerning manufacture of the vaccine and quality control testing (see Section C.2.1 and C.2.2) should be submitted to the authorities. This information shall be provided from three consecutive vaccine batches with a volume not less than 1/3 of the typical industrial batch volume.

2.3.2. Safety requirements

All vaccines must be safety tested. Safety tests for attenuated vaccines do not address the issue of their teratogenicity. Attenuated virus vaccines are teratogenic and should not be administered to pregnant sheep during the first half of pregnancy as this may cause fetal abnormalities and embryo death (Flanagan & Johnson, 1995; Lunt *et al.*, 2006).

i) Target and non-target animal safety

Demonstration of avirulence is necessary for live, attenuated vaccines. A number of sheep, seronegative by an appropriate, sensitive serological test (that will reliably detect antibodies even in vaccinated animals), are inoculated with the primary seed stock. Temperatures are noted twice daily. The animals are monitored at regular intervals over a period of 28 days for clinical signs and any local or systemic reactions to ensure avirulence and innocuity. Blood samples removed at regular intervals can be used to measure level of viraemia and antibody responses. The test shall be valid if all of the vaccinated sheep show evidence of virus replication and do not display signs of disease other than mild transient illness. In South Africa, a clinical reaction index is calculated for each animal between days 4 and 14 and must be below a specific standard value.

ii) Reversion-to-virulence for attenuated/live vaccines and environmental considerations

Transmissibility is an issue with live attenuated vaccines but not with killed vaccines. Procedures to determine if attenuated virus can be transmitted by insects that feed on vaccinated, viraemic sheep are difficult to perform and analyse statistically, and consequently, this criterion of vaccine validation is rarely sought. Laboratory data indicate that laboratory-adapted viruses can be transmitted by insect vectors (Ferrari *et al.*, 2005; Monaco *et al.*, 2006; Standfast *et al.*, 1985). A suitable procedure to determine attenuated virus transmissibility requires that sheep be vaccinated and, during viraemia, that they be exposed to competent, uninfected *Culicoides*, which are then permitted to feed on uninfected animals that are monitored for the presence of BTV and anti-BTV antibody. As the titre of attenuated virus in the blood of vaccinated sheep is usually low, very large numbers of *Culicoides* may be needed and only a small proportion of these would become infected and live long enough to feed on and potentially transmit the virus to other uninfected sheep. It is difficult to design a laboratory experiment that takes account of the large numbers of vaccinated sheep and insects that would be present in field situations. Although virus titres in blood less than 10^3 TCID₅₀/ml have traditionally been considered a "safe" threshold, authentic instances of insects acquiring BTV from animals with viraemic titres much less than 10^3 TCID₅₀/ml have been reported (Bonneau *et al.*, 2002). Given the complex interaction of BTV, *Culicoides* vectors and animal hosts in the life cycle of infection, virus titres induced by live attenuated vaccine should be kept to an absolute minimum especially if field transmission of vaccine strains is a concern.

Current data indicate that during viraemia and in contrast to wild-type virus, laboratory-adapted strains of BTV may be found in the semen of bulls and rams (Kirkland *et al.*, 2004). The implications of these observations for virus transmissibility are unclear. A recent study of semen from rams vaccinated with BTV2 live attenuated vaccine showed that even if BTV was not detected in the semen, the vaccine caused a decrease in the quality of the semen (Bréard *et al.*, 2007).

Validation studies confirm that attenuated viruses do not revert to virulence in vaccinated sheep. However, if attenuated viruses can be transmitted from vaccinated animals, reversion to virulence during a number of sheep-insect replication cycles becomes a distinct prospect. The only appropriate way to monitor for reversion to virulence under

these circumstances is to compare the virulence of the vaccine virus with that which had been subject to several sheep–insect replication cycles as described above. As indicated, this is difficult to achieve. Consequently, the effect of a number of sheep–insect passages on the virulence of attenuated viruses has not been determined. In South Africa, it is accepted that if blood from vaccinated animals during the viraemic stages is serially passaged three times in sheep without reversion to virulence, the chances of reversion in the field will be infinitely small. In Europe, five passages are required.

iii) Precautions (hazards)

Attenuated vaccines should be used in the cooler months when adult *Culicoides* vector populations are at a minimum. They should not be used in ewes during the first half of pregnancy and in rams 2 months before the breeding season

2.3.3. Efficacy requirements

Vaccinated and unvaccinated sheep known to be susceptible to BT disease should be challenged with virulent homologous serotype. It is recommended that the challenge model use preferably virus passaged only in ruminant animals and with no or limited ECE or cell culture passages. Passage in such an isolation system results in viral cultures that might induce clinical BT disease that is milder than the natural disease (Flanagan & Johnson, 1995). Animals are monitored for clinical signs of BT, rectal temperatures are taken twice daily and blood samples removed at regular intervals to measure viraemia and antibody responses. Unvaccinated control sheep should show clinical signs of BT and viraemia. However, it is difficult to be certain of the appearance of clinical disease following inoculation of sheep with certain BTV serotypes and isolates, and consequently, evidence of infection of unvaccinated control sheep may rest on the appearance of a temperature rise above 40°C and a viraemia. As a further evidence of infection pre- and post-vaccination sera are checked for the presence of neutralising antibody.

2.3.4. Vaccines permitting a DIVA strategy (detection of infection in vaccinated animals)

The live attenuated and inactivated products now commercially available don't allow any DIVA strategy.

2.3.5. Duration of immunity

Studies to determine a minimum duration of immunity should be conducted before the vaccine receives final approval. Duration of immunity should be demonstrated in a manner similar to the original efficacy study, challenging animals at the end of the claimed period of protection. At a minimum, the duration should be for the length of the mosquito season in seasonally infected areas. It may be desirable to demonstrate longer immunity for animals at higher risk and in infected areas with year-round mosquito activity.

2.3.6. Stability

Live and inactivated vaccines are typically assigned an initial dating of 18 or 24 months, respectively, before expiry. Real-time stability studies should be conducted to confirm the appropriateness of all expiration dating. Product labelling should specify proper storage conditions.

REFERENCES

- AFSHAR A., THOMAS F.C., WRIGHT P.F., SHAPIRO J.L. & ANDERSON J. (1989). Comparison of competitive ELISA, indirect ELISA and standard AGID tests for detecting bluetongue virus antibodies in cattle and sheep. *Vet. Rec.*, **124**, 136–141.
- ATTOUI H., MERTENS P.P.C., BECNEL J., BELAGANAHALLI S., BERGOIN M., BRUSSAARD C.P., CHAPPELL J.D., CIARLET M., DEL VAS M., DERMODY T.S., DORMITZER P.R., DUNCAN R., FANG Q., GRAHAM R., GUGLIELMI K.M., HARDING R.M., HILLMAN B., MAKKAY A., MARZACHI C., MATTHIJNSSENS J., MILNE R.G., MOHDJAAFAR F., MORI H., NOORDELOOS A.A., OMURA T., PATTON J.T., RAO S., MAAN M., STOLTZ D., SUZUKI N., UPADHYAYA N.M., WEI C. & ZHOU H. (2012). Family reoviridae. *In: Virus Taxonomy: Classification and Nomenclature of Viruses: Ninth Report of the International Committee on Taxonomy of Viruses*, King A.M.Q., Adams M.J., Carstens E.B. & Lefkowitz, E.J., eds. Elsevier Academic Press, San Diego, USA.
- BATTEN C.A., BACHANEK-BANKOWSKA K., BIN-TARIF A., KGOSANA L., SWAIN A.J., CORTEYN M., DARPEL K., MELLOR P.S., ELLIOTT H.G. & OURA C.A.L. (2008). Bluetongue virus: European Community inter-laboratory comparison tests to evaluate ELISA and RT-PCR detection methods. *Vet. Microbiol.*, **129**, 80–88.

- BRÉARD E., POZZI N., SAILLEAU C., CATINOT V., DURAND B., DUMONT P., GUÉRIEN B. & ZIENTARA S. (2007). Transient effect of the attenuated bluetongue virus vaccine on the quality of the ram semen. *Vet. Rec.*, **160**, 431–435.
- BONNEAU K.R., DEMAULA C.D., MULLENS B.A. & MACLACHLAN N.J. (2002) Duration of viraemia infectious to *Culicoides sonorensis* in bluetongue virus-infected cattle and sheep. *Vet. Microbiol.*, **88**, 115–125.
- CLAVIJO A., HECKERT R.A., DULAC G.C. & AFSHAR A. (2000). Isolation and identification of bluetongue virus. *J. Virol. Methods*, **87**, 13–23.
- DANIELS P.W., SENDOW I., PRITCHARD L.I., SUKARSIH & EATON B.T. (2004). Regional overview of bluetongue viruses in South-East Asia: viruses, vectors and surveillance. *Veterinaria Italiana*, **40**, 94–100.
- DUNGU B., GERDES T. & SMIT T. (2004). The use of vaccination in the control of bluetongue in southern Africa. *Vet. Ital.*, **40**, 616–622.
- ERASMUS B.J. (1975). The control of bluetongue in an enzootic situation. *Aust. Vet. J.*, **51**, 209–210.
- FLANAGAN M. & JOHNSON S.J. (1995). The effects of vaccination of Merino ewes with an attenuated Australian bluetongue virus serotype 23 at different stages of gestation. *Aust. Vet. J.*, **72**, 455–457.
- FERRARI G., DE LIBERATO C., SCAVIA G., LORENZETTI R., ZINI M., FARINA F., MAGLIANO A., CARDATI G., SHOLL F., GUIDONI M., SCICLUNA M.T., AMADDEO D., SCARAMOZZINO P. & AUTORINO G.L. (2005). Active circulation of bluetongue vaccine virus serotype-2 among unvaccinated cattle in central Italy. *Prev. Vet. Med.*, **68**, 10–13.
- GARD G.P., WEIR R.P. & WALSH S.J. (1988). Arboviruses recovered from sentinel cattle using several isolation methods. *Vet. Microbiol.*, **18**, 119–125.
- GIBBS E.P. & GREINER E.C. (1994). The epidemiology of bluetongue. *Comp. Immunol. Microbiol. Infect. Dis.*, **17**, 207–220. Review.
- GOULD A.R. (1987). The complete nucleotide sequence of bluetongue virus serotype 1 RNA3 and a comparison with other geographic serotypes from Australia, South Africa and the United States of America, and with other orbivirus isolates. *Virus Res.*, **7**, 169–183.
- HOFMANN M., GRIOT C., CHAIGNAT V., PERLER L. & THÜR B. (2008). Bluetongue disease reaches Switzerland. *Schweiz. Arch. Tierheilk.*, **150**, 49–56.
- JOHNSON D.J., WILSON W.C. & PAUL P.S. (2000). Validation of a reverse transcriptase multiplex PCR test for the serotype determination of U.S. isolates of bluetongue virus. *Vet. Microbiol.*, **76**, 105–115.
- KATZ J., GUSTAFSON G., ALSTAD D., ADLER K. & MOSER K. (1993). Colorimetric diagnosis of prolonged bluetongue viremia in sheep using an enzyme-linked oligonucleotide sorbent assay of amplified viral nucleic acids. *Am. J. Vet. Res.*, **54**, 2021–2026.
- KIRKLAND P.D., MELVILLE L.F., HUNT N.T., WILLIAMS C.F. & DAVIS R.J. (2004) Excretion of bluetongue virus in cattle semen: a feature of laboratory adapted virus. *Vet. Ital.*, **40**, 497–501.
- KRAMPS J.A., VAN MAANEN K., MARS M.H., POPMA J.K. & VAN RIJN P.A. (2008). Validation of a commercial ELISA for the detection of bluetongue virus (BTV)-specific antibodies in individual milk samples of Dutch dairy cows *Vet. Microbiol.*, **130**, 80–87.
- LUNT R.A., MELVILLE L., HUNT N., DAVIS S., ROOTES C.L., NEWBERRY K.M., PRITCHARD L.I., MIDDLETON D., BINGHAM J., DANIELS P.W. & EATON B.T. (2006). Cultured skin fibroblast cells derived from bluetongue virus-inoculated sheep and field-infected cattle are not a source of late and protracted recoverable virus. *J. Gen. Virol.*, **87**, 3661–3666.
- LUNT R.A., WHITE J.R. & BLACKSELL S.D. (1988). Evaluation of a monoclonal antibody blocking ELISA for the detection of group-specific antibodies to bluetongue virus in experimental and field sera. *J. Gen. Virol.*, **69**, 2729–2740.
- MAAN N., MAAN S., BELAGANAHALLI M., OSTLUND E., JOHNSON D., NOMIKOU K. & MERTENS P. (2012). Identification and differentiation of the twenty-six bluetongue virus serotypes by RT-PCR amplification of the serotype-specific genome segment 2. *PLoS One*, **7** (2): e32601. Doi: 10.1371/journal.pone.0032601.
- MACLACHLAN N.J. (2004). Bluetongue: pathogenesis and duration of viraemia. *Veterinaria Italiana*, **40**, 462–467.
- MACLACHLAN N.J., NUNAMAKER R.A., KATZ J.B., SAWYER M.M., AKITA G.Y., OSBURN B.I. & TABERCHNICK W.J. (1994). Detection of bluetongue virus in the blood of inoculated calves: comparison of virus isolation, PCR assay, and *in vitro* feeding of *Culicoides variipennis*. *Arch. Virol.*, **136**, 1–8.
- MACLACHLAN N.J., OSBURN B.I., STOTT J.L. & GHALIB H.W. (1985). Orbivirus infection of the bovine fetus. *Prog. Clin. Biol. Res.*, **178**, 79–84.

- MARTYN C.J., GOULD A.R. & EATON B.T. (1990). High level expression of the major core protein VP7 and the non-structural protein NS3 of bluetongue virus in yeast: use of expressed VP7 as a diagnostic, group-reactive antigen in a blocking ELISA. *Virus Res.*, **18**, 165–178.
- MCCOLL K.A. & GOULD A.R. (1991). Detection and characterisation of bluetongue virus using the polymerase chain reaction. *Virus Res.*, **21**, 19–34.
- MCHOLLAND L.E. & MECHAM J.O. (2003). Characterization of cell lines developed from field populations of *Culicoides sonorensis* (Diptera: Ceratopogonidae). *J. Med. Entomol.*, **40**, 348–351.
- MERTENS P.P., MAAN N.S., PRASAD G., SAMUEL A.R., SHAW A.E., POTGIETER A.C., ANTHONY S.J. & MAAN S. (2007). Design of primers and use of RT-PCR assays for typing European bluetongue virus isolates: differentiation of field and vaccine strains *J. Gen. Virol.*, **88**, 2811–2823.
- MERTENS P.P.C., MAAN S., SAMUEL A. & ATTOUI H. (2005). Orbiviruses. *In: Virus Taxonomy. Classification and Nomenclature of Viruses*; Fauquet C.M., Mayo M.A., Maniloff J., Desselberger U. & Ball L.A., eds., Elsevier Academic Press, Amsterdam, Netherlands, 466–483.
- MONACO F., CAMMÀ C., SERINI S. & SAVINI G. (2006). Differentiation between field and vaccine strain of bluetongue virus serotype 16. *Vet. Microbiol.*, **116**, 45–52.
- OLDFIELD S., ADACHI A., URAKAWA T., HIRASAWA T. & ROY P. (1990). Purification and characterization of the major group-specific core antigen VP7 of bluetongue virus synthesized by a recombinant baculovirus. *J. Gen. Virol.*, **71**, 2649–2656.
- OSBURN B.I. (2004). Conclusions. *In: Proceedings of the Third OIE International Symposium on Bluetongue*. Taormina, 26–29 October 2003, MacLachlan N.J. & Pearson J.E., eds. *Vet. Ital.*, **40**, 713–726.
- POTGIETER A.C., MONACO F., MANGANA O., NOMIKOU K., YADIN H. & SAVINI G. (2005). VP2-segment sequence analysis of some isolates of bluetongue virus recovered in the Mediterranean basin during the 1998–2003 outbreak. *J. Vet. Med. [B.]*, **52**, 372–379.
- PRITCHARD L.I., GOULD A.R., WILSON W.C., THOMPSON L., MERTENS P.P. & WADE-EVANS A.M. (1995). Complete nucleotide sequence of RNA segment 3 of bluetongue virus serotype 2 (Ona-A). Phylogenetic analyses reveal the probable origin and relationship with other orbiviruses. *Virus Res.*, **35**, 247–261.
- STANDFAST H.A., DYCE A.L. & MULLER M.J. (1985). Vectors of BT in Australia. *In: Bluetongue and Related Orbiviruses*, Barber T.L. & Jochim M.M., eds. Alan R. Liss, New York, USA, 177–186.
- VENTER G.J., GERDES G.H., MELLOR P.S. & PAWESKA J.T. (2004). Transmission potential of South African *Culicoides* species for live-attenuated bluetongue virus. *Vet. Ital.*, **40**, 198–202.
- VERWOERD D.W. & ERASMUS B.J. (2004). Bluetongue. *In: Infectious Diseases of Livestock*, Second Edition, Coetzer J.A.W. & Tustin R.C., eds. Oxford University Press Southern Africa, Cape Town, South Africa, 1201–1220.
- WECHSLER S.J., MCHOLLAND L.E. & TABACHNICK W.J. (1989). Cell lines from *Culicoides variipennis* (Diptera: Ceratopogonidae) support replication of bluetongue virus. *J. Invert. Pathol.*, **54**, 385–393.
- WILSON W.C., MA, H.C., VENTER E.H., VAN DJIK A.A., SEAL B.S. & MECHAM J.O. (2000). Phylogenetic relationships of bluetongue viruses based on gene S7. *Virus Res.*, **67**, 141–151.

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NB: There are OIE Reference Laboratories for Bluetongue
(see Table in Part 4 of this *Terrestrial Manual* or consult the OIE Web site for the most up-to-date list:
<http://www.oie.int/en/scientific-expertise/reference-laboratories/list-of-laboratories/>).

Please contact the OIE Reference Laboratories for any further information on
diagnostic tests, reagents and vaccines for bluetongue

NB: FIRST ADOPTED IN 1991; MOST RECENT UPDATES ADOPTED IN 2014.