Influence of the uterine environment on the development of in vitro-produced equine embryos

Katrien Smits, Jan Govaere, Luc J Peelman¹, Karen Goossens¹, Dirk C de Graaf², Dries Vercauteren³, Leen Vandaele, Maarten Hoogewijs, Eline Wydooghe, Tom Stout⁴ and Ann Van Soom

Department of Reproduction, Obstetrics and Herd Health, Faculty of Veterinary Medicine, Ghent University, Salisburylaan 133, B-9820 Merelbeke, Belgium, ¹Department of Nutrition, Genetics and Ethology, Faculty of Veterinary Medicine, Ghent University, Heidestraat 19, 9820 Merelbeke, Belgium, ²Laboratory of Zoophysiology, Ghent University, K.L. Ledeganckstraat 35, 9000 Gent, Belgium, ³Department of Pharmaceutics, Ghent University, Harelbekestraat 72, 9000 Gent, Belgium and ⁴Department of Equine Sciences, Faculty of Veterinary Medicine, Utrecht University, Yalelaan 114, 3584CM Utrecht, The Netherlands

Correspondence should be addressed to K Smits; Email: katrien.smits@ugent.be

Abstract

The necessity for early interaction between the embryo and the oviductal and/or uterine environment in the horse is reflected by several striking differences between equine embryos that develop in vivo and those produced in vitro. Better understanding of the salient interactions may help to improve the efficiency of in vitro equine embryo production. In an initial experiment, cleavage-stage in vitro-produced (IVP) equine embryos were transferred into the uterus of recipient mares that had ovulated recently to determine whether premature placement in this in vivo environment would improve subsequent development. In a second experiment, an important element of the uterine environment was mimicked by adding uterocalin, a major component of the endometrial secretions during early pregnancy, to the culture medium. Intrauterine transfer of cleavage-stage IVP equine embryos yielded neither ultrasonographically detectable pregnancies nor day 7 blastocysts, indicating that the uterus is not a suitable environment for pre-compact morula stage horse embryos. By contrast, exposure to uterocalin during IVP improved capsule formation, although it did not measurably affect the development or expression of a panel of genes known to differ between in vivo and in vitro embryos. Further studies are required to evaluate whether uterocalin serves purely as a carrier protein or more directly promotes improved capsule development.

Introduction

Early embryonic development in the horse is characterised by a number of peculiarities (Betteridge 2007). First, the equine embryo does not exit the oviduct via the prominent uterotubal papilla until as late as 144–156 h after ovulation (Freeman et al. 1991). Moreover, unfertilised eggs are not capable of stimulating passage through the ampullary–isthmic junction and are instead retained in the oviduct. The stimulus to this selective oviductal transport is the stage-specific production of prostaglandin E₂ by days 4–5 equine conceptuses (Weber et al. 1991), and is a clear example of very early embryo-maternal interaction in the horse.

Another enigmatic feature exemplifying early embryo-maternal interaction in the horse is the formation of an acellular glycoprotein tertiary embryo coat (‘capsule’: Flood et al. 1982) very soon after the arrival of the horse embryo in the uterus; the capsule completely envelopes the equine conceptus until around day 21 of gestation (Betteridge 2007). Although the precise functions of the capsule are not clear, it has been proposed to function as a ‘mailbox’ that collects endometrial components such as signalling molecules and nutrients and transports them to the embryo (Herrler & Beier 2000), and to maintain the spherical shape of the mobile embryo and physically protect the while it migrates around the uterus to signal its presence to its dam and prevent luteolysis (Allen & Stewart 2001, Stout & Allen 2001). In addition, structural changes in the capsule are thought to be instrumental to the processes of fixation and orientation of the conceptus within the mare’s uterus (Oriol 1994).

Since a functional chorioallantoic placenta is not formed until as late as days 40–45 of gestation, equids have the longest pre-implantation period of all mammals studied to date (Allen & Stewart 2001); moreover, during this prolonged pre-implantation period, the embryo is entirely dependent on endometrial secretions

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(histotrophe) for its nutrition. An undoubtedly important component of this histotrophe is the 19 kDa progesterone-dependent protein, uterocalin, which is secreted by the endometrial glands during both dioestrus and early pregnancy (Crossett et al. 1996). The marked drop in uterocalin production that coincides with the disappearance of the capsule at around day 21 and the fact that uterocalin is one of the most abundant proteins in the capsule (Quinn et al. 2007) suggest that there may be a functional correlation between uterocalin production and capsule persistence (Crossett et al. 1996). Moreover, detection of uterocalin in the trophoblast and yolk sac fluid of equine conceptuses implies passage through the capsule and absorption by the conceptus proper. On the basis of its structure, uterocalin has been classified as a member of the lipocalin family, which contains several transport proteins that bind small hydrophobic molecules (Crossett et al. 1996). Moreover, in-depth structural analysis of uterocalin suggests a putative function as a carrier of essential lipids and amino acids for the developing conceptus (Kennedy 2004).

All of the above illustrate the importance of embryo-maternal interaction during early embryonic development in the horse. Furthermore, when equine embryos are produced in vitro, and therefore in the absence of the maternal tract, they differ markedly from their in vivo counterparts in terms of the kinetics of development, incidence of apoptotic cells, inner cell mass morphology and gene expression patterns (Tremerola et al. 2003, Pomar et al. 2005, Smits et al. 2011). One of the more striking irregularities of in vitro-produced (IVP) horse embryos is the failure of normal capsule formation (Tremerola et al. 2003); even though capsular mucin-like glycoproteins are produced in vitro, they fail to coalesce into the distinct continuous capsule observed around in vivo equine embryos from the early blastocyst stage. The reason(s) for the failure of capsular glycoprotein coalescence in vitro are not known, but may involve simple dispersion of the glycoproteins into the culture medium, thereby preventing attainment of the critical concentration required for capsule assembly, or failure of hydration and cross-linking of the capsular glycoproteins in the absence of a specific uterine component(s) (Tremerola et al. 2003). In either case, the presence of the mare’s uterus appears to be essential to the process of capsule formation. To confirm that the uterine environment and/or specific uterine components are central to capsule formation, we exposed IVP embryos either to the complete uterine environment or to the endometrial protein, uterocalin, which is known to contribute substantially to the capsule of day 10–18 blastocysts. A recent study demonstrated that temporary transfer of IVP day 7 horse blastocysts to the mare’s uterus for 2–3 days had a positive effect on capsule formation, as assessed by light microscopy (Choi et al. 2009). In this study, we wanted to further determine whether ‘premature’ transfer of day 2–3 embryos to the uterus would enhance capsule formation and improve equine blastocyst development rates and quality, compared with culture in vitro. Earlier studies involving intrauterine transfer of small numbers of in vivo-derived horse embryos were not very successful (Allen & Rowson 1975, Ball et al. 1989, Weber et al. 1993), but ‘premature intrauterine transfer’ with large numbers of day 2–3 IVP horse embryos has not been described previously. In this latter respect, while it is common practice to culture embryos to the blastocyst stage before intrauterine transfer in most domestic species, in human medicine, premature intrauterine transfer of day 2 and 3 IVP embryos is a routine procedure that yields good results (Younis et al. 2009) and circumvents the potential downsides of prolonged in vitro culture or the difficulty of transferring early embryos to the oviduct. Currently, blastocyst rates of 25–35% can be obtained after ICSI and in vitro culture of cleaved horse embryos (Hinrichs 2010). However, only few laboratories are able to obtain such good results. If transfer of cleavage-stage IVP embryos to the uterus of the mare was successful, it would considerably simplify IVP, even if it did not have additional beneficial effects on embryonic development and capsule formation. To more specifically investigate the putative role of uterocalin in capsule formation and early development of equine embryos, recombinant uterocalin was added to the culture medium for 5–10 days and the effect on subsequent development was examined in terms of capsule formation and expression of a panel of genes known to be differentially expressed by in vivo vs IVP horse embryos.

Results

Experiment 1: intrauterine transfer of cleavage-stage IVP embryos

A total of 99 cleaved (2–8 cell stage) embryos were transferred to the uterus of 12 synchronised (days 2–3 after ovulation) mares (average of 8.25 embryos/mare). Six of these mares were subsequently examined for pregnancy by transrectal ultrasound on day 14 after ovulation, but no conceptus vesicles were detected. The uterus of the remaining six mares was flushed on day 7 after ovulation. Disappointingly, embryos were recovered from only three of the six mares and no mare yielded more than a single embryo (overall recovery rate 6%). Moreover, none of the three recovered embryos had developed to the blastocyst stage; instead all three were clearly degenerate.

Experiment 2: addition of recombinant uterocalin to embryo culture medium

No significant differences in overall development were observed between embryos that had or had not been exposed to uterocalin during IVP (Table 1). In total, 60%
Table 1 Influence of uterocalin on capsule formation and development of in vitro-produced equine blastocysts (Equus caballus).

<table>
<thead>
<tr>
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<th>Control</th>
<th>Uterocalin</th>
<th>P value</th>
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<tr>
<td>Blastocyst percentage (%)</td>
<td>12.6 (±2.5)</td>
<td>13.3 (±3.5)</td>
<td>0.84</td>
</tr>
<tr>
<td>Mean cell count D9.5 (×1000)</td>
<td>551 (±47)</td>
<td>579 (±40)</td>
<td>0.66</td>
</tr>
<tr>
<td>Mean diameter (μm)</td>
<td>270 (±12)</td>
<td>247 (±15)</td>
<td>0.25</td>
</tr>
<tr>
<td>OC-1 specific fluorescence</td>
<td>2123 (±117)</td>
<td>2745 (±208)</td>
<td>0.03</td>
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Addition of uterocalin during IVC did not affect blastocyst development rate, embryo cell number or diameter. Total immunofluorescence of embryonic capsular glycoproteins was significantly increased by exposure to uterocalin. Mean values and their respective S.E.M. are shown.

The five genes analysed by quantitative real-time PCR (RT-qPCR), BEX2, FABP3, HSP90AA1, MOBKL3 and ODC, were chosen as markers for developmental quality because a previous study indicated downregulation of these genes in IVP compared with in vivo-derived equine blastocysts (Smits et al. 2011). It was hypothesised that adding the endometrial protein, uterocalin, might induce an expression pattern in the in vitro embryos more closely resembling that of in vivo embryos. In fact, no differences in expression levels were found between the blastocysts which were cultured with uterocalin (n=11) and the blastocysts from the control group (n=11) (Fig. 4). In this experiment, RT-qPCR efficiencies of ≥99% and correlation coefficients of ≥0.992 were obtained, indicating that the results were reliable.

Discussion

The ability to efficiently produce equine embryos in vitro is of interest for both research and the clinical treatment of (equine) infertility. Currently, the acceptable rates of production of viable embryos can be obtained using ICSI of in vitro-matured oocytes followed by culture in DMEM/F12 supplemented with serum; however, there is still room for further optimisation of the in vitro culture process to improve overall efficiency (Galli et al. 2007, Hinrichs et al. 2007, Blanco et al. 2009). Understanding the influence of the equine oviductal and uterine environments, and specific components thereof, on
early embryonic development could lead to targeted adaptations of the in vitro environment to mimic aspects of the maternal environment identified as beneficial.

In this study, intrauterine transfer of cleavage-stage IVP embryos did not yield any pregnancies and, since no healthy blastocysts were recovered on day 7 (i.e. 4–5 days after transfer), it appears that the transferred cleavage-stage embryos do not develop in the uterine environment. There are several possible explanations for this failure of development. Firstly, it is possible that closure of the cervix 2–3 days after ovulation was suboptimal, and allowed loss of the embryos via the cervix. However, pregnancy after transfer of day 10 embryos to recipient mares on day 1 or 3 after ovulation has been described by Wilsher et al. (2010) and, although none of the embryos subsequently developed into normal pregnancies (i.e. with an embryo proper), it does illustrate that the uterus should be mechanically capable of retaining embryos introduced soon after ovulation. Wilsher et al.’s (2010) study also illustrates the possibilities and limits of embryo-uterine synchrony. Optimal synchronisation between donor and recipient mares yields the best results; it is, however, not known at which stage after ovulation the uterus of a recipient mare is best suited to nourishing embryonic stages that normally develop in the oviduct, especially when development may have been delayed by production in vitro. In addition, the large numbers of transferred embryos could have had a negative effect. In vivo, there are rarely more than two to three embryos in a mare’s uterus. The choice to transfer an average of more than eight embryos per mare was based on recipient availability. Even if development of some embryos might have been impeded, recovery of others by flushing on day 7 would have been expected. Another possible explanation is simple absence of intrinsic developmental potential of the transferred in vitro embryos. However, using identical preliminary steps, standard in vitro production yielded 5–10% blastocysts in our hands, and transfer of one of these blastocysts resulted in pregnancy and the birth of a live foal (Smits et al. 2010); in short, at least some (5–10%) of the cleaved embryos should have been capable of further development.

The remaining possible interpretation is that the mare’s uterus does not provide an adequate or appropriate environment for early cleavage-stage embryos. A similar failure to establish pregnancy following premature intrauterine transfer was reported for two mouse embryos of 292 days (Goto et al. 1993), and in previous small studies that described the intrauterine transfer of early in vivo derived horse embryos. For example, Weber et al. (1993) achieved no pregnancies following intrauterine transfer of two horse embryos of 7 days and, while Allen & Rowson (1975) did describe a single pregnancy after transfer of three equine embryos of 5 days, the age of the embryo was estimated from daily examination for ovulation by transrectal palpation; the embryo that resulted in pregnancy could thus easily have been closer to 4 days. Indeed, day 4 equine embryos have been reported to be sufficiently mature to survive in the mare’s uterus (Peyrot et al. 1987).

In human medicine, cleavage-stage embryos are routinely transferred to the uterus and, while the procedure is considered to entail both specific advantages and disadvantages compared with blastocyst transfer, overall favourable pregnancy rates of 30–40% are common (Bromer & Seli 2008, Papanikolaou et al. 2008). The reason for the marked differences in the success of intrauterine transfer of day 2–3 embryos in women and some primates compared with other domestic species might be the anatomical differences. In the mare, there is a distinct uterotubal papilla which presumably helps to maintain the marked differences in
may be a maternally derived structural component of the capsule rather than just a transiently associated transport molecule. Previous studies have demonstrated that OC-1-reactive capsular glycoproteins are secreted by trophectoderm cells (Albihn et al. 2003), while the failure of normal capsule formation in vitro suggests the need for an additional maternal component (Tremoleda et al. 2003). Indeed, the 19 kDa endometrial protein, uterocalin, was first isolated by SDS-PAGE as one of the dominant proteins released by equine embryonic capsules (Stewart et al. 1995). Subsequent studies confirmed the presence of uterocalin in the capsule in a temporal pattern, which suggested a functional correlation between the two (Crossett et al. 1996, Herrler & Beier 2000, Quinn et al. 2007). As a result of its molecular structure, uterocalin has since been proposed to function primarily as a carrier of biologically important lipids and a source of essential amino acids for the developing conceptus (Suire et al. 2001, Kennedy 2004), where the positive charge of uterocalin (Crossett et al. 1996) is thought to facilitate its binding to the negatively charged sialic acid residues of the capsule (Oriol et al. 1993). Since uterocalin has also been found in trophoblast cells (Crossett et al. 1996, Ellenberger et al. 2008), some of the molecule clearly passes through the capsule and presumably fulfils a role as a carrier protein. This study suggests that uterocalin also contributes to the structure of the capsule and may play a role in the initial aggregation and cross-linking of trophectoderm-produced OC-1-reactive glycoproteins. In summary, uterocalin appears to be instrumental in initial capsular glycoprotein coalescence, contributes to the structure of the capsule and plays an important role in transporting essential nutrients and/or signalling molecules to the early intrauterine conceptus.

The dynamics of embryonic covering formation and loss and, in particular, the addition of tubal- and uterine-secreted materials during development has been described in several species (for review see Denker 2000). In this respect, the equine embryonic capsule has been proposed to be most analogous to the neozona of the rabbit (Bettridge 1989, Herrler & Beier 2000). For example, while trophectodermal secretions are important for neozona formation, maternal components also appear to be critical (Fischer et al. 1991, Denker 2000). Indeed, in vitro culture of rabbit embryos is not only associated with the deposition of granular material on the inside of the mucoprotein layer, but also with failure of normal neozona formation, aberrant herniation of embryonic cells through the zona, and incomplete or failed dissolution of the zona pellucida (Fischer et al. 1991). These features are reminiscent of the aberrations of capsule formation and hatching observed in IVP equine embryos (Tremoleda et al. 2003, Stout et al. 2005). However, in both species, these deviations are not necessarily irreversible and most can be corrected by exposure to uterine components. In this respect, addition
of uterine flushings to rabbit embryo culture medium, or intrauterine transfer of \textit{in vitro}-cultured embryos, has been shown to allow reactivation and completion of zona dissolution, although uterine flushings alone did not induce neo zona formation \textit{in vitro} (Fischer et al. 1991). In the horse, intrauterine transfer of IVP blastocysts results in successful pregnancies (Galli et al. 2007, Hinrichs et al. 2007, Smits et al. 2010) with an apparently normal capsule (Choi et al. 2009), illustrating that the degree of initial disturbance to capsule formation during IVP is not so severe as to preclude normal pregnancy, at least as long as exposure to the uterine environment is sufficiently early to remedy the aberrations; by contrast, total removal of the blastocyst capsule from day 6.5 embryos is incompatible with embryonic survival after transfer (Stout et al. 2005); clearly, the disruption to capsule formation suffered during IVP is not equivalent to removal.

In this study, some aspects of aberrant equine embryonic capsule formation \textit{in vitro} appeared to be ameliorated by uterocalin. However, it is not known how uterocalin had this effect, and neither was uterocalin alone sufficient to completely normalise capsule formation; e.g. the ‘hatched’ areas of trophoectoderm still exhibited dispersed patches of OC-1-reactive glyco-proteins that did not become confluent (Fig. 2). In addition, capsular material still penetrated into the transzonal channels (Fig. 1) as previously reported for IVP embryos (Tremoleda et al. 2003), but not seen around \textit{in vivo} embryos. While it is possible that there was insufficient uterocalin provided to completely normalise capsule production, it is more likely that this partial correction reflects the continued absence of other components of the complex intrauterine environment that are required for capsule formation.

Surprisingly, no clear influence of uterocalin on the development of equine IVP blastocysts was observed, as illustrated by equal blastocyst formation rates, embryo diameters and cell counts between control and uterocalin groups (Table 1). Moreover, exposure to uterocalin did not alter the expression of five genes (\textit{HSP90AA1}, \textit{MOBKL3} and \textit{ODC}; Fig. 4) previously found to be downregulated in IVP embryos compared with \textit{in vivo}-derived embryos (Smits et al. 2011). A similar failure of endometrial proteins to influence early equine embryonic development was reported by Bogh et al. (2002) who exposed day 8 \textit{in vivo} derived embryos to a p19 (i.e. uterocalin) homologue during a 3-h incubation, and found no obvious influence on subsequent embryonic growth and metabolism. The absence of effects on embryonic growth, metabolism (Bogh et al. 2002), gene expression or quality (this study) might be an artefact due to measurement of factors not influenced by uterocalin. However, it is also possible that a crucial ligand(s) necessary for uterocalin to influence gene expression or metabolism was absent in the \textit{in vitro} culture medium; this would seem logical if uterocalin is primarily a carrier protein, as suggested by its ability to bind several essential small lipids (Suire et al. 2001), rather than a stimulator of embryonic growth or development per se.

**Conclusion**

Several unusual features of early embryonic development in the horse illustrate the importance of embryo–maternal interaction. To optimise equine IVP, it may well be necessary to further clarify, and develop methods to mimic, particular aspects of this interaction. In this study, an essential role of the oviductal environment was illustrated by the failure of premature intrauterine transfer to yield viable embryos. In addition, the need for contact with the uterine environment of slightly older embryos was illustrated by the positive effect on capsule development of exposure to the maternal endometrial protein, uterocalin. However, since capsule formation in the presence of uterocalin was not completely normal, while embryo development and gene expression still differed markedly from \textit{in vivo} embryos, it is clear that many aspects of the maternal environment necessary to support optimal early embryonic development still need to be identified.

**Materials and Methods**

**Experiment 1: intrauterine transfer of cleavage-stage equine in vitro embryos**

All animal procedures were approved by the ethics committee of the Faculty of Veterinary Medicine at Ghent University. \textit{In vitro} embryos were produced as previously described by Smits et al. (2010). Briefly, slaughterhouse oocytes were matured \textit{in vitro} for 24 h in a DMEM/F12-based medium in an atmosphere containing 5% CO$_2$. MII oocytes were fertilised by conventional ICSI and cultured \textit{in vitro} in DMEM/F12 with 10% FCS at 38.5 °C in 5% CO$_2$, 5% O$_2$ and 90% N$_2$. On day 2–3 after ICSI, the cleaved embryos were transferred by means of a transcervical pipette (IMV Technologies France, L’Aigle, France) to the uterus of a recipient mare that had ovulated 2–3 days previously. A total of 99 cleaved embryos were transferred to the uterus of 12 synchronised mares (average of 8.25 embryos per mare). The transfers were performed by an experienced veterinarian, who obtained average pregnancy rates of 85% on day 14 and 79% on day 45 when using grades 1 and 2 day 7 horse blastocysts. On the day of transfer, the recipient mare was injected intravenously with 1.1 mg/kg of the nonsteroidal anti-inflammatory agent, flunixin meglumine (Embolin, Emdoka Bvda, Hoogstraten, Belgium); in addition, daily \textit{per os} treatment with 0.044 mg/kg of the synthetic prostogestogen, altrenogest (Regumate, Intervet, Boxmeer, The Netherlands) was initiated and continued until evaluation for embryo development. Half of the mares were examined by \textit{per rectum} uterine ultrasound 14 days after ICSI, and the other half were subjected to embryo recovery by transcervical uterine lavage.
as described by McKinnon & Squires (2009), on day 7 after ICSI. Briefly, flushing was performed with 6 l of lactated Ringer’s solution introduced and recovered using a Bivona catheter (Minitüb, Tiefenbach, Germany); the recovered fluid was passed through an EZ filter (Bioniche Pharma, Galway, Ireland). Any embryos recovered were stained with Hoechst 33342 (Invitrogen) to assess cell viability and number.

**Experiment 2: IVP in the presence of recombinant uterocalin**

*Production of recombinant uterocalin*

A recombinant uterocalin clone and an anti-uterocalin–antibody were kindly provided by Prof. M W Kennedy (University of Glasgow, UK). The recombinant uterocalin was purified mainly as described by Suire et al. (2001) using the Profinity IMAC Ni-Charged Resin (BioRad) to produce a working concentration of 7.88 mg/ml.

**Estimation of the physiological concentration of uterocalin**

Since no absolute concentrations of uterocalin in the uterine environment are reported in the literature, the physiological concentration of uterocalin was estimated using a uterine secretion sample recovered from a day 7 pregnant mare. Sampling of uterine secretions was performed by means of aspiration through a pipette for deep intrauterine insemination as described by Velazquez et al. (2010), while subsequent uterine lavage resulted in the recovery of an embryo, thereby confirming that the mare had been pregnant. A dot blot technique was used to compare a dilution series of recombinant uterocalin with the recovered uterine secretion and indicated that the concentration of uterocalin in the uterine secretions was ~4 mg/ml. Subsequent analysis of the uterine secretions of three other pregnant mares revealed an average concentration of 0.8 mg/ml. This physiological uterocalin concentration was mimicked in the experiment by 1 mg/ml recombinant uterocalin.

**IVP of equine blastocysts**

*In vitro* embryos were produced as described for experiment 1, except that only oocytes with a compact cumulus complex were used, the maturation time was 28 h and ICSI was performed by a Piezo Drill (Prime Tech Ltd., Ibaraki, Japan). The embryos were cultured in groups of 10–20 in 20 µl droplets of DMEM/F12 with 10% FCS at 38.5 °C in 5% CO₂, 5% O₂ and 90% N₂. On day 2.5, half of the medium was refreshed and the embryos that had not cleaved were removed. On day 6, half of the medium was refreshed again and, in half of the culture droplets, 2.54 µl of the medium was replaced by the recombinant uterocalin solution, resulting in a final concentration of 1 mg/ml recombinant uterocalin. On day 9–9.5, the embryos that had reached the blastocyst stage were recovered for further analysis.

**Immunofluorescent staining of the capsule**

Immunofluorescent staining of the equine capsule was performed as described by Tremoleda et al. (2003) using the monoclonal anti-capsule antibody OC-1 (Oriol et al. 1993), which was kindly provided by Prof. K J Betteridge (University of Guelph, Canada). Day 9.5 blastocysts were fixed in 4% paraformaldehyde (P6118; Sigma–Aldrich) and stored at 4 °C until analysis. Twelve blastocysts that had been cultured with uterocalin and 14 blastocysts from the control group were stained simultaneously. After permeabilisation by exposure to 0.5% (v/v) Triton X-100 for 30 min at room temperature, the blastocysts were washed three times in phosphate buffered saline (PBS) containing 1 mg/ml polyvinylpyrrolidone (PVP). Non-specific staining was blocked by incubation in 10% (v/v) goat serum (16210-064; Invitrogen) for 30 min at 37 °C. The blastocysts were then washed again and incubated with the primary antibody (mouse monoclonal anti-capsule OC-1: 1/200 dilution) for 1.5 h at 37 °C. In both groups, a negative control blastocyst was incubated in 10% goat serum without primary antibody. After a washing step, incubation with the secondary antibody (goat anti-mouse FITC; Invitrogen), 1/100 dilution) was performed for 1 h at 37 °C, followed by another washing step. Nuclei were then stained by incubation with 2% propidium iodide (Invitrogen) for 30 min at room temperature, after which the embryos were fixed in Dabco (Thermo Fisher Scientific, Geel, Belgium) on siliconised glass and enclosed under a coverslip supported by small vaseline bridges to prevent crushing of the embryos. All embryos were evaluated in one session using a Nikon C1 confocal laser scanning module attached to a motorised Nikon TE2000-E inverted microscope (Nikon BeLux, Brussels, Belgium) and identical settings. Subsequent fluorescence measurements were performed by Nikon EC-V1 FreeViewer software. Since some blastocysts were slightly squeezed by the coverslip, intact capsules of six uterocalin blastocysts and eight control blastocysts in similar condition were evaluated. For each embryo, the total fluorescence of three areas in different, randomly selected spots of the capsule was measured and the mean of these three measurements was recorded (Fig. 5).

**RT-qPCR**

For both the uterocalin and the control groups, 11 blastocysts were selected on day 9. After washing in Dulbecco’s PBS (DPBS),
individual blastocysts were transferred to cryotubes with 2 μl lysis buffer, frozen in liquid nitrogen for 3 min and stored at −80 °C. RNA extraction was performed by the RNeasy Micro Kit (Qiagen) and, after RT minus control, the RNA was converted into cDNA using the iScript cDNA synthesis Kit (BioRad). The expression of the five development ‘marker’ genes (BEX2, FABP3, HSP90AA1, MOBKL3 and ODC) was quantified by RT-qPCR as described by Smits et al. (2011). Normalisation of data was performed by UBC, ACTB, RPL32 and GAPDH as reference genes (Smits et al. 2009).

**Statistical analysis**

The blastocyst development rates for the uterocalin and control groups were compared using a Pearson χ² test (SPSS 16.0; SPSS, Inc., Chicago, IL, USA). Cell number and capsular fluorescence were compared between the groups by -tests, while gene expression between the uterocalin and control groups was compared with a Mann–Whitney U test using GraphPad InStat 3 (GraphPad Software Inc, La Jolla, CA, USA).

**Declaration of interest**

The authors declare that there is no conflict of interest that could be perceived as prejudicing the impartiality of the reported research.

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