Complex expression patterns support potential roles for maternally derived activins in the establishment of pregnancy in mouse

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Abstract

Maternal–fetal communications are critical for the establishment of pregnancy. Embryonic growth and differentiation factors produced by the oviduct and uterus play essential roles during the pre- and early post-implantation phases. Although several studies indicate roles for activin in embryonic development, gene-knockout studies have failed to identify a critical role in mammalian embryogenesis. We hypothesized that activin is produced by maternal tissues during the establishment of pregnancy, and thus maternally derived activin could compensate for the absence of embryonic activin in null homozygotes during critical developmental stages. We investigated the expression of inhibin \( \alpha \), activin \( \beta A \), and \( \beta B \) subunits in the mouse oviduct and uterus during the estrous cycle and early pregnancy, and in the early conceptus. Inhibin \( \alpha \) subunit was weakly expressed, while activin \( \beta A \) and \( \beta B \) subunits were strongly expressed in oviduct and uterus at estrous, and dramatically upregulated in the uterus on each day of pregnancy between days 3.5 and 8.5 post coitum. Prior to implantation, activin \( \beta A \) and \( \beta B \) subunits were immunolocalized to oviductal and uterine epithelial cells; following implantation they were expressed in the stroma, in a wave preceding decidualization. Later in pregnancy, activin \( \beta A \) and \( \beta B \) subunits were present in decidua basalis, trophoblast giant cells, and labyrinth zone of the developing placenta. Expression of activin \( \beta A \) subunit was also detected in blastocysts and early post-implantation embryos. These data are consistent with a role for maternally derived activins in the support of the pre-implantation embryo, and during gastrulation and embryogenesis.


Introduction

Embryo development occurs in a highly specialized maternal environment within the oviduct and uterine lumen during the pre-implantation phases, and subsequently within the uterine implantation site. Cytokines and growth factors secreted by maternal cells support embryo development, as demonstrated by an improved developmental rate when blastocysts are co-cultured in vitro with endometrial and oviductal cells (Seta 2001). Although cytokines, such as granulocyte macrophage colony-stimulating factor and leukemia inhibitor factor, are reported to facilitate mouse embryo development (Robertson & Seamark 1992, Kauma & Matt 1995, Tsai et al. 2000), many maternally derived factors remain to be identified. This is critical in order to understand the regulation of early post-implantation embryo development, and improve culture conditions to more closely mimic the maternal environment for embryo culture following in vitro fertilization (IVF).

Activins are potential candidate factors for maternal–embryo interactions, due to their roles in regulating cell proliferation, differentiation and apoptosis, and their abundant expression and actions in remodeling tissues. They are abundantly expressed by reproductive tissues, including the uterus, and have been attributed with roles in decidualization, embryogenesis, and organogenesis in a variety of species (Jones et al. 2002).

Activins are dimeric glycoproteins belonging to the transforming growth factor-\( \beta \) (TGF-\( \beta \)) superfamily. Activin A is a dimer of \( \beta A \) subunits, while activin B is formed from \( \beta B \) subunits. These two subunits can also heterodimerize to form activin AB (\( \beta A: \beta B \)), or alternatively can dimerize with
a distinct α subunit to form inhibin A (α;βA) or inhibin B (α;βB). Activins A and B are believed to have similar actions, with differences in specific functions ascribed to distinct spatial and temporal expressions. Inhibins are potent inhibitors of activins, acting as competitive antagonists through activin receptors (Harrison et al. 2005). Activins bind to cell-surface type II receptors, ActRIIA or ActRIIB, which then recruit the type I receptor, ActRI/ALK-4. This initiates a signaling cascade through serine/threonine kinase activity, and the phosphorylation and nuclear translocation of intracellular Smads.

Studies in both mice and humans provide evidence for roles for activins during the establishment of pregnancy. Mouse, bovine, and human embryos express activin receptors during the pre-implantation phase (Lu et al. 1993, Yoshioka et al. 1998, He et al. 1999). Receptor expression (ActRI/ALK-4, ActRII, and ActRIIB) is maintained in the post-implantation embryo, and during organogenesis, receptors are expressed in a tissue-specific manner (Feijen et al. 1994, Manova et al. 1995, Verschueren et al. 1995, Roberts et al. 1996). Moreover, pre-implantation mouse and human embryos express activin βA and βB subunits; at the blastocyst stage, they become localized to the inner cell mass (Albano et al. 1993, Lu et al. 1993, He et al. 1999). The expression of activin β subunit in the early post-implantation phase is controversial (van den Eijnden-van Raaij et al. 1992, Manova et al. 1995), but strong expression is detected from E8.5 to 10.5 onwards (Feijen et al. 1994, Roberts & Barth 1994).

Despite the demonstration that activin A is obligatory for mesoderm induction in Xenopus (Smith et al. 1990), gene manipulation studies in the mouse have failed to demonstrate an absolute requirement for activins in mammalian embryogenesis (Vassalli et al. 1994, Matzuk et al. 1995). This discrepancy could be attributed either to compensatory actions of multiple activin β subunits or to maternal supply of activins during pregnancy. As activin βA null mice die neonatally, it has not been possible to establish whether embryo development is adversely affected by the absence of maternal activin. In support of the latter, in vitro studies with mouse and bovine embryos indicate that exogenous activin A promotes morula and blastocyst development (Orimo et al. 1996, Yoshioka et al. 1998, Mtango et al. 2003).

In humans, activin βA and βB subunits are abundantly expressed by uterine epithelial glands and decidual cells during the peri-implantation phase and in early pregnancy (Leung et al. 1998, Otani et al. 1998, Jones et al. 2000). Furthermore, activin A is secreted into the uterine fluid that supports pre-implantation development (Petraglia et al. 1998). Throughout the first trimester of pregnancy, endometrial glandular products, including activin A, are secreted into the intervillus space and thus have sustained roles in supporting embryonic and placental development (Hempstock et al. 2004). Throughout pregnancy, inhibin and activin subunits are also abundantly produced by the placenta, and have been implicated in the regulation of placental development and hormone production (Petraglia et al. 1989, Caniggia et al. 1997).

Expression of activin βA subunits by the decidua in rat and mouse in the early post-implantation phase has been reported (Manova et al. 1992, Feijen et al. 1994, Gu & Gibori 1995). However, maternal production of inhibin/activin subunits during the pre-implantation phase, and in the implantation site in the mouse, has not been characterized, leaving major gaps in our understanding of potential maternal–embryo communication through the activin system. We hypothesize that maternally derived activins play critical roles in promoting pre- and early post-implantation embryo development, and subsequently in the differentiation events and tissue remodeling during uterine decidualization and placentation.

We report here the expression and production of inhibin α, βA, and βB in the uterus across the mouse estrous cycle and during the establishment of pregnancy. Spatial/temporal expression, and relative abundance of βA and βB subunits were examined to indicate whether there are overlapping or distinct functions. These data provide evidence consistent with a potential role for maternally derived activin during embryo development/implantation, and verify that the mouse would be a suitable model for functional studies to provide a greater understanding of activin involvement in human implantation.

Materials and Methods

Animals

Non-pregnant and pregnant Swiss White out-bred mice (Mus musculus) were used (Animal Services, Monash University). Mice were housed and handled according to the Monash University animal ethics guidelines on the care and use of laboratory animals. Experimental procedures were approved by the Monash University Animal Ethics Committee (Committee B, Monash Medical Centre, Melbourne, Australia) for animal experimentation and conducted in accordance with the Australian National Health and Medical Research Council (NHMRC) guidelines.

The stage of the estrous cycle of non-pregnant mice (8–10 weeks, weight 20–22 g) was determined by analysis of vaginal smears. Stage of cycle was determined by the characteristic presence or absence of inflammatory leukocytes, and the histological appearance of epithelial cells. Mice were killed by CO₂ asphyxiation and cervical dislocation during the proestrous, estrous, metestrous, and diestrous phases (n = 2/stage), and oviducts and uteri dissected. Ovaries were collected from randomly cycling mice to use as positive controls.
Female mice (8–10 weeks, weight 20–22 g) were mated with male mice, and checked for vaginal plugs at 0900 h the following morning. Mice were killed on days 3.5, 4.5, 5.5, 6.5, 8.5, 10.5, and 12.5 post coitum (pc); where day 0 is morning of vaginal plug and uteri dissected (n=2/day of pregnancy). On day 3.5 pc, ovaries were examined for corpora lutea to confirm ovulation. On day 4.5 pc, implantation sites were identified by tail vein injection of Chicago blue dye (1% in saline, 0.1 ml/mouse), 5 min before killing. Uterine tissues were dissected out immediately and processed for immunohistochemistry and RNA extraction. Implantation and inter-implantation sites were collected separately on days 4.5–6.5 of pregnancy; thereafter only implantation sites were collected. On day 10.5, the implantation site was dissected to separate the embryo from the implantation site.

Samples were divided into two portions: for immunohistochemistry, tissues were immersion fixed in formalin overnight at 4°C, prior to thorough washing in Tris-buffered saline (TBS, pH 7.6) and paraffin embedding; the remaining samples were snap-frozen for subsequent RNA extraction.

**In vitro fertilization and embryo culture**

**Oocytes**

Six-week-old females were superovulated by an i.p. injection of 10 IU equine chorionic gonadotropin (Folligon, Intervet International, Boxmeer, The Netherlands) followed by 10 IU hCG (Chorulon, Intervet International, The Netherlands) in 0.2 ml saline 50 h later. At 13–14 h after hCG injection, oocyte–cumulus complexes were collected from the oviducts by incubation for 5 min in HEPES-KSOMaa (Biggers et al. 2000; BDH Chemicals Pty., Ltd, Melbourne, Victoria, Australia) containing 60 IU/ml hyaluronidase (Sigma type IV-S; Sigma Chemicals Co.). Adhering cumulus cells were removed from oocytes by gentle pipetting and washed in HEPES-KSOMaa. Oocytes were then placed into 20 μl drops of modified KSOMaa previously equilibrated under mineral oil in 5% CO2 in air at 37°C. Oocytes were incubated for 1 h at 37°C in 5% CO2 in air before they were used for IVF.

**Sperm**

Sperm were collected from 10- to 12-week-old males. Briefly, both the caudae epididymides were dissected from the mice and a small slat was made in each cauda before transfer to 2 ml modified Tyrode’s medium (Fraser 1984; BDH) and equilibrated in 5% CO2 in air at 37°C in a 35 mm culture dish (NUNC, Roskilde, Denmark) under oil. The tissue was then removed from the dish 1 h later, and the sperm concentration was examined and adjusted to 3–5×10⁶ sperm/ml. Sperm were capacitated for a total of 2 h before IVF.

In vitro fertilization (IVF)

For IVF, cumulus-free oocytes (30–50) were incubated for 5 h, under mineral oil, in 2 ml capacitated sperm solution (3–5×10⁶ sperm/ml). Oocytes were removed from the sperm solution after 5 h, washed in HEPES-KSOMaa, and cultured at 37°C in modified KSOMaa under mineral oil in 5% CO2 in air. Six hours later, oocytes were examined for signs of normal fertilization. Oocytes with two pronuclei and a second polar body were regarded as fertilized. These oocytes were separated from unfertilized oocytes and cultured in modified KSOMaa at 37°C under 5% CO2 in air up to 96 h for development to the blastocyst stage in vitro. Two and four cell embryos, morulae, and blastocysts (unhatched; n=3/stage) were collected after 24, 48, 56–60, and 72 h culture respectively, placed directly into 100 μl TRIZOL reagent (Invitrogen, Mt Waverly, Victoria, Australia). Samples were then snap-frozen prior to RNA extraction.

**RNA extraction and purification**

Total RNA was extracted from frozen uterine samples by acid guanidinium thiocyanate–phenol–chloroform extraction (GTC; Sigma) as previously described (Nie et al. 2000), followed by purification through RNAeasy spin columns (Qiagen) according to the manufacturer’s instructions. RNA was extracted from oviducts using RNAeasy spin columns. RNA samples were treated with RNase-free DNase I (Ambion, Austin, Texas, USA) to remove genomic DNA contamination, and analyzed by spectrophotometry to determine RNA concentration, yield, and purity. RNA concentrations were also analyzed by Ribogreen fluorescence RNA assay (Invitrogen) as previously described (Jones et al. 2004).

RNA was isolated from embryos frozen in TRIZOL using a modified protocol. Samples were allowed to thaw and incubated at room temperature for 30 min, followed by vortexing. Thereafter, the manufacturer’s instructions were followed, with the exception of the inclusion of a second chloroform – only extraction to prevent phenol carryover. RNA samples were resuspended in 12 μl diethylpyrocarbonate (DEPC) H2O, and the entire volume was utilized for RT.

**Real-time RT-PCR**

Reverse transcription and validation

Total RNA (1 μg) was reverse transcribed in triplicate reactions using random hexameric primers and AMV-RTase (Roche), at 46°C for 90 min. Omission of reverse transcriptase served as a negative control. Triplicate RTs were analyzed for efficiency and reproducibility of the RT reaction by real-time PCR for ribosomal 18S subunit using a Light Cycler (Roche) as previously described (Jones et al. 2004). Primers used were: sense 5’-CGG CTA CCA CAT AAC CGA CCA CAG CTG G-3’; antisense 5’-GGA ATT TGG GCC TGA ATT AAG CTC C-3’. Primers were designed for a 200 bp fragment using Primer Express software (Applied Biosystems, Foster City, CA). Real-time PCR was performed on the Light Cycler (Roche) under the following conditions: 1 cycle at 95°C for 10 min, primers at 95°C for 10 sec, 60°C for 5 sec and 72°C for 10 sec for 40 cycles, followed by melting curve analysis. The Light Cycler was controlled by a Thermal Cycler software (Roche) for data acquisition. Samples were examined for both the lowest Ct and the highest efficiency to ensure optimal amplification. A dissociation curve was generated following each real-time PCR reaction to test for non-specific products and primer dimers. The Light Cycler data were analyzed by the software to calculate the relative amount of cDNA. An increase of 0.05 is considered significant and accepted as a difference. Results are expressed as mean ± standard error of the mean.
CCA AGG AA-3' and antisense 5'-GCT GGA ATT ACC GCG GCT-3'. Amplified DNA quantitation was performed using SYBR Green I, by comparison with serially diluted cDNA standards, generated by PCR as previously described (Jones et al. 2004). Cycle conditions were as follows: 95 °C for 10 min, 35 cycles of 95 °C for 15 s, 60 °C for 5 s, and 72 °C for 10 s. 18S concentrations (pg/μl) were compared between triplicates and the intra-assay variability of the RT and PCR steps was evaluated. Within triplicates, samples outside 15% variability of the average 18S concentration were excluded as outliers. Otherwise, triplicates were pooled, creating cDNA reaction products that are close representations of the initial mRNA population. Negative controls were performed by omission of RT. All PCR products were DNA sequenced to confirm identity.

Real-time PCR for inhibin/activin subunits

Pooled RT samples (diluted in the ratio of 1:10) were amplified in a Light Cycler for inhibin/activin subunits, using primers: inhibin α: 5'-GCC TTG GCC TTG TGC TCC TGC G-3'/-ACG CGT AGG GAG GTC ATG CTC C-3'; activin βA: 5'-GCC TTG AGT GCG ACG GC-3'/ 5'-GCA GCC ACA CTC CTC-3'; activin βB: 5'-TCT TCA TCG ACT TTC GCC TCA T-3'/-TGT CAG GCG C AG C CAC TCC T-3' (Sigma; Albano et al. 1993). Cycle conditions were as follows: 95 °C for 10 min, 40 cycles of 95 °C for 15 s, 55/62/60 °C for 5 s, and 72 °C for 15 s. PCRs were run in duplicate or triplicate. Mouse ovarian cDNA, diluted in the ratio of 1:5, was included in each run as a positive and quality control.

Immunohistochemistry for inhibin/activin subunits

Inhibin/activin α, βA, and βB subunits were immunolocalized using affinity purified rabbit polyclonal antibodies raised against the individual inhibin subunits (anti-porcine inhibin α1–26, anti-porcine inhibin βA81–113, and anti-human inhibin βB86–112; kind gift from Prof. Wylie Vale, La Jolla, CA, USA; Vaughan et al. 1989), as previously described (Jones et al. 2000), but with modifications for optimal staining of mouse tissues. Mouse ovary was included as a positive control. In brief, microwave antigen retrieval was performed (5 min HIGH, 5 min LOW in a 1 kW microwave oven, followed by 20-min cooling), and endogenous peroxidase activity blocked with aqueous H2O2. Primary antibodies were applied at 2 μg/ml in a non-immune block containing 20% goat serum and 0.1% Tween-20 in TBS. Negative controls were performed by substitution of primary antibody with 2 μg/ml rabbit IgG (X0936; Dako, Glostrup, Denmark). Antibody binding was thereby detected by sequential application of conjugate (Dako) for 1 h at room temperature, with chromogen diaminobenzidine (DAB; Dako), forming an insoluble brown precipitate. Tissue sections were counterstained with Harris’s hematoxylin, dehydrated, and mounted with DPX mounting medium (Sigma).

Results

Expression of inhibin/activin α, βA, and βB subunits in uterus in non-pregnant cycling mice

A positive signal was obtained for all primer pairs for ovarian RT samples included as a positive control in real-time PCRs (data not shown). No amplification product was detected in negative controls. Inhibin α subunit mRNA was almost undetectable in cycling uterine samples (Fig. 1A). In contrast, activin βA mRNA was abundantly expressed in non-pregnant uterine samples, with fluctuations across the estrous cycle. mRNA levels were increased greater than threefold from the proestrous to estrous phases, thereafter levels decreased in the metestrous phase (Fig. 1A). A similar pattern was detected for βB subunit mRNA (Fig. 1A). Moreover, both activin β subunits were expressed by the oviduct, with an identical cyclical expression pattern, and maximal upregulation observed in estrous phase (data not shown).

Immunohistochemistry verified this expression pattern at the protein level and identified the cellular location. Intense immunostaining was detected for α, βA, and βB subunits in ovarian follicles and corpora lutea in agreement with known established patterns reviewed by Findlay (1993) (Fig. 1B and C). βA and βB subunits immunolocalized to epithelial cells lining the oviduct (Fig. 1D), and in the uterus, to the luminal and glandular epithelium (Fig. 1E–L). Consistent with mRNA expression patterns, endometrial immunostaining levels varied markedly during the estrous cycle. In the proestrous phase, βA subunit was present in the luminal epithelium (Fig. 1E). In the estrous phase, immunostaining levels were increased in luminal and glandular epithelial cells (Fig. 1F), and in addition, intensely stained single cells (possibly infiltrating immune cells) were identified in the sub-epithelial zone (Fig. 1G). Following estrous, βA immunostaining was minimal or absent in metestrous (Fig. 1H) and diestrous phases (data not shown). βB subunit protein expression was low in proestrus endometrium in both the luminal and the glandular epithelium (data not shown), but was elevated in the estrous phase in all epithelial cells (Fig. 1I). No intensely stained stromal cell was detected in the sub-epithelial zone (Fig. 1J). In metestrous (Fig. 1K) and diestrous (Fig. 1L) endometria, βB immunostaining was severely diminished in luminal epithelial cells, but retained in epithelial glands during both post-estrus phases. Low immunostaining was detected for α subunit in oviduct (Fig. 1B), whereas none was detectable in the uterus (Fig. 1M).

Expression of activin subunits in mouse pre-implantation embryos

Real-time PCR was conducted for activins βA and βB on embryos at the two cell, four cell, morula, and blastocyst
(unhatched) stages \((n=3\) pooled embryo RNA/stage). A positive signal for both activin \(\beta A\) and \(\beta B\) subunits was detected in ovary included as a positive control (12.8 fg \(\beta A\) mRNA/pg 18S, 4.25 fg \(\beta B\) mRNA/pg 18S). \(\beta A\) mRNA was below the limit of detection in all embryos except for blastocysts (3.5 fg \(\beta A\) mRNA/pg 18S); \(\beta B\) expression was not detectable in embryos, including blastocysts (Fig. 2). 18S was used as a control for RNA integrity.

**Expression of inhibin/activin \(\alpha\), \(\beta A\), and \(\beta B\) subunits during establishment of pregnancy**

Inhibin \(\alpha\) mRNA was undetectable by real-time PCR in the uterus in the peri-implantation phase (days 3.5–4.5 pc; Fig. 3A). Very low levels of expression were detected in the implantation site from days 5.5 to 8.5. In contrast, activin \(\beta A\) mRNA was markedly upregulated on day 3.5 pc compared with non-pregnant (NP) uteri (greater than tenfold; Fig. 3B, inset). On the day of embryo attachment (day 4.5 pc), activin \(\beta A\) was increased in one out of two implantation site samples, while no change in mRNA expression was seen in inter-implantation sites. From day 5.5, activin \(\beta A\) subunit expression rose further, with expression levels approximately tenfold higher in both implantation and inter-implantation sites compared with day 4.5 (Fig. 3B). Despite considerable variation, reflecting the heterogeneity of this dynamic tissue, a further increase (approximately threefold) in \(\beta A\) mRNA
expression was detected on each of the days 6.5 and 8.5 pc in implantation sites, while levels remained static in inter-implantation sites. At day 10.5, expression levels were examined in the uterus and embryo separately. This showed a sharp decline in uterine activin βA mRNA expression and low embryonic expression of βA subunit. A similar expression pattern was detected for βB mRNA; however, expression levels were consistently 50–100 times lower than βA mRNA levels (Fig. 3C). Furthermore, the increase in βB subunit expression with the onset of pregnancy was delayed compared with that of βA, with only a slight increase in activin βB mRNA from non-pregnant to day 4.5 (Fig. 3C, inset). However, on day 5.5, βB mRNA was upregulated more than 30-fold in implantation sites, and fivefold in inter-implantation sites, compared with day 4.5 (Fig. 3C). Expression increased a further threefold in implantation sites on day 6.5 and thereafter a slight decline was detected on days 8.5–10.5 in the uterine tissues. Activin βB mRNA expression was detected in the embryo, but at very low levels compared with activin βA mRNA expression.

**Immunohistochemistry for inhibin/activin subunits**

Consistent with mRNA expression patterns, βA protein was upregulated on day 3.5 of pregnancy, in luminal and glandular epithelium (Fig. 4A) and also in the stromal zone underlying the luminal epithelium, particularly in the anti-mesometrial region. With blastocyst attachment on day 4.5, stromal immunostaining became more intense and polarized at the anti-mesometrial zone, where the primary decidual response occurs (Fig. 4B and C). Immunostaining for βA was downregulated in the luminal epithelium in implantation site compared with NP. A distinct staining pattern was observed throughout inter-implantation sites, with intense immunostaining in epithelial glands, while luminal epithelium and stroma possessed minimal immunoreactivity (Fig. 4D). By day 5.5 pc, activin βA expression exhibited a ‘butterfly-like’ pattern, with intense immunoreactivity in the anti-mesometrial and mesometrial zones distant to the implanting embryo, and with stroma immediately surrounding the embryo devoid of immunostaining (Fig. 4E and F). Basal epithelial glands maintained strong activin βA expression (Fig. 4E). Between days 6.5 and 8.5 pc, uterine immunoreactivity for activin βA diminished and became limited to the basal zone (bz) adjacent to the myometrium (Fig. 4G and J). From day 5.5 onwards, intense immunostaining for activin βA was observed in and surrounding the developing embryonic and extra-embryonic structures (Fig. 4F, H and I).

An almost identical pattern of activin βB was observed in the implantation site (Fig. 5). Intense staining was detected in epithelial glands throughout early pregnancy
Figure 4 Immunohistochemical localization of activin βA in mouse uterus during early pregnancy. Photomicrographs are orientated such that the mesometrium is at the lower right-hand side in each case. (A) Day 3.5 post coitum (pc), (B) and (C) day 4.5 showing blastocyst (collapsed) attachment to the anti-mesometrial pole and activin βA staining in the primary decidual zone (pdz), (D) day 5.5 pc inter-implantation, and (E) day 5.5 pc implantation site, showing staining in the embryo (em) and epithelial glands (gl), (F) high magnification of embryo at day 5.5 pc, (G) day 6.5 pc implantation site, illustrating the restriction of activin βA staining to the embryo and basal zone (bz) of the decidua, (H) high magnification of embryo at day 6.5 pc, (I) activin βA immunostaining in the embryo and developing placenta, (J) anti-mesometrial zone showing staining restricted to the basal zone, and faint staining in giant cells (gc). Scale bars = 100 μm, except (B, E, G) = 200 μm and (F, H) = 50 μm.
(e.g., Fig. 5A and G). Activin βB immunostaining was faint or absent in the embryo in the early post-implantation phases (Fig. 5C–F), but it intensely immunolocalized to individual maternal cells surrounding the implanting embryo on day 5.5 (Fig. 5D), and was generally present in the mesometrial zone surrounding the developing extra-embryonic structures on days 6.6–8.5 (Fig. 5E and F).

From days 8.5 to 12.5 pc, strong staining for activin βA (Fig. 6A) and βB (Figs 5H and 6C) subunits was detected in the mesometrial decidua basalis, localized specifically to decidual cells, while uNK cells were unstained (Fig. 6B). During this time, immunoreactivity for activin subunits was also detected in the developing placenta. Inhibin α immunostaining was faint or absent in the developing placenta (Fig. 6D). Immunostaining for both activin subunits was present in enlarged giant cells (Fig. 6E) and in the trophoblast cells of the labyrinth zone (Fig. 6A and F). Activin βA was absent from the spongiotrophoblast layer (Fig. 6A and G).
while βB immunostaining was present, but at reduced levels compared with decidua and labyrinth (Fig. 6C and H). Of interest was the intense staining for βB in the cells lining the large maternal vessels of the spongiotrophoblast layer (Fig. 6H).

**Discussion**

Activin βA and βB subunits were found to be strongly expressed in the mouse female reproductive tract during the estrous cycle and establishment of pregnancy. In non-pregnant cycling mice, activin β subunit mRNA, and

![Figure 6](https://www.reproduction-online.org) Immunohistochemical localization of inhibin/activin subunits in mouse placenta on day 12.5 post coitum (pc). (A) Activin βA immunoreactivity is intense in labyrinth (lab) and decidua basalis (dec), but faint or absent in spongiotrophoblast (sp), (B) high magnification images of βA decidual cells, (C) activin βB positively immunostained labyrinth, spongiotrophoblast, and decidua, while (D) inhibin α immunostaining was absent from all placental layers, (E) activin βA staining in trophoblast giant cells, and (F) activin βB staining in placental labyrinth, (G) activin βA is absent from spongiotrophoblast, while (H) activin βB is present around blood vessels (v) in the spongiotrophoblast. Scale bars on (A, C and D) = 200 μm, on (H) = 100 μm, and on (B, E–G) = 50 μm.
protein expression levels fluctuated in a cycle-dependent manner in the uterus. During early pregnancy, both activin β subunits were strikingly upregulated in uterine tissues, and embryonic activin βA mRNA expression was detectable from the blastocyst stage. Immunohistochemical studies localized activin βA and βB subunits to the uterine epithelial and decidual cells, as well as to the developing embryo and placenta.

The predominant source of activin β subunits during the estrous cycle and pre-implantation phase were the epithelial cells lining the oviduct and endometrium. The strong expression of activin subunits by the oviductal epithelial cells is consistent with findings by Lu et al. (1993) and with a role for maternally derived activins, secreted into oviductal fluid, in facilitating pre-implantation embryo development. Both βA and βB subunits mRNA and protein were elevated at estrus, suggesting positive regulation by estrogen, and a ‘priming’ of the oviduct and uterus in anticipation of pregnancy. A further increase in activin βA expression occurred in the uterus on day 3.5, corresponding to the entry of morulae into the uterine lumen and the first embryonic differentiation steps of morula compaction and blastocyst development, hatching and implantation.

The elevation in maternally derived activin correlating these critical steps is consistent with an involvement for activin in facilitating embryonic development. Furthermore, embryonic expression of activin βA mRNA becomes detectable with blastocyst development. ActRs are expressed by the embryo during this period, and in the human, expression levels of both type I and II receptors increase with blastocyst formation (He et al. 1999). Indeed, treatment of mouse and bovine embryos with activin A in vitro accelerates morula and blastocyst development, blastocyst hatching, and increases blastocyst cell number (Orimo et al. 1996, Yoshioka et al. 1998, Mtango et al. 2003); while neutralization of endogenous activin with follistatin treatment in vitro significantly retards embryonic development (Yoshioka et al. 1998). Although activin action is not obligatory for pre-implantation development, as embryos lacking ActRs survive beyond the blastocyst stage (Gu et al. 1998, Song et al. 1999), it is likely that the deficit of maternal growth factors is responsible for the retarded development observed during embryo culture (Seta 2001). Therefore, the corroboration between the previous in vitro data and the specific temporal expression patterns for activin subunits in oviductal and uterine epithelia, support the hypothesis that activins contribute to the maternal support of pre-implantation embryo development.

The upregulation of activin βA and βB subunit expressions in the implantation site in early pregnancy is primarily due to expression by the developing decidua, and later, by the placental cells. Activin βA expression has been well characterized during decidualization in the rat (Gu et al. 1995), and here we demonstrate a similar expression pattern for both βA and βB in the mouse decidua. Importantly, the upregulation of activin subunits in stromal cells precedes decidualization, with a coordinated wave of up- and downregulation spreading through the uterus in advance of the onset and spread of decidualization. This indicates a preparative role for activin prior to decidual differentiation. In human endometrial stromal cells, activin upregulates matrix metalloproteinases (MMPs; Jones et al. 2006), which have critical actions during decidualization, at least in the rat (Alexander et al. 1996, Rechtman et al. 1999). Interestingly, in the rodent uterus, MMP-2 and -3 have almost identical expression patterns to that of activin during early pregnancy (Alexander et al. 1996, Bany et al. 2000), suggesting an interaction between activins and MMPs during decidualization in the mouse. Later in pregnancy, activin promotes decidual apoptotic regression in the rat, allowing expansion of the implantation site for placentation and embryonic growth (Gu et al. 1995, Tessier et al. 2003). Interestingly, the primary decidual cells adjacent to the embryo on day 7.5 that stain intensively for activin subunits correspond to the site of maximum apoptosis (Alexander et al. 1996).

In the developing placenta, activin β subunit expression was evident in the invasive trophoblast giant cells at the maternal–fetal interface, possibly related with a role in promoting trophoblast invasion, as has been demonstrated in the human (Caniggia et al. 1997). In the placenta at day 12.5, activin βA and βB subunits were clearly localized predominantly to the syncytiotrophoblast of the labyrinth. This is identical with the spatial expression of activin/ inhibin subunits in the villous syncytiotrophoblast of the human placenta (Lockwood et al. 1997); their expression is apparent immediately with the onset of syncytialization both in vivo and in vitro (Debieve et al. 2000, Jones et al. 2006).

The significance of multiple activin β subunits is not known. βA and βB subunits frequently exhibit distinct expression patterns, suggesting specific functions. For example, in developing embryos, widespread expression of βA in mesenchymally derived tissues, including gonads and brain, has been reported (Manova et al. 1992); while βB expression is primarily limited to reproductive tissues. However, in the uterus, βA and βB have almost identical expression patterns, but βB mRNA is consistently far less abundant than βA. This may explain the failure of previous studies to detect βB subunit in uterine and embryonic tissues using less sensitive methods (Manova et al. 1992). Female mice lacking activin βB subunit undergo normal implantation and early pregnancy, but experience perinatal loss of progeny, suggesting maternal activin B fulfils critical functions late in pregnancy (Vassalli et al. 1994). Fertility cannot be assessed in the βA knockout, due to neonatal lethality (Matzuk et al. 1995). However, when the βB subunit was ‘knocked-in’ into the βA gene locus of the βA-null mutant, the female mice reproduced, but
exhibited a severely decreased pregnancy rate (Brown et al. 2000). The uterine activin βA subunit deficiency contribution to reduced fertility has not been established. However, these gene manipulation models suggest distinct functions for βA and βB in the female reproductive system.

The present study provides evidence for the uterine epithelial (glandular) and decidual cells as a significant source of activin in the mouse at the time of implantation and early embryonic development, the uterine epithelial (glandular) and decidual cells are a significant source of activin in the mouse. Therefore, during gastrulation, when activin action is obligatory in Xenopus, lack of embryonic βA and βB could be compensated for by the abundant maternal contribution, giving a plausible explanation for the absence of gastrulation-related defects in the activin βA null (−/−) embryos (in activin βA subunit heterozygote (+/−) mothers). This is supported by the absolute requirement for activin signaling for mesoderm induction in mice, with arrested development prior to gastrulation (et al. 1999). The results of the present study strongly support the hypothesis that maternally derived activin fulfills a critical role in early embryonic development in the mouse.

In summary, we have characterized in detail the spatial and temporal expression patterns of inhibin/activin α, βA, and βB subunits during the establishment of pregnancy in the mouse. We demonstrate that maternally derived activin subunits, predominantly βA, are specifically upregulated in the uterine luminal epithelium during pre-implantation embryo development, and subsequently in the decidual of the implantation site, during the critical development events of gastrulation and organogenesis. These data suggest that activins secreted by the oviduct and uterus could facilitate the development of the embryo prior to implantation. Furthermore, our findings support the hypothesis that maternally derived activins play important roles in early post-implantation embryo development. The observed expression patterns in the mouse uterus bear marked resemblance to those in the human uterus during the preparation for implantation and in early pregnancy, thus suggesting it to be a suitable model to delineate the functional roles for activins in maternal–fetal interactions during the establishment of pregnancy.

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