

## Article

# Human Adipose CD34<sup>+</sup>CD90<sup>+</sup> Stem Cells and collagen scaffold constructs grafted *in vivo* fabricate loose connective and adipose tissues<sup>†</sup>

Giuseppe A. Ferraro<sup>1\*</sup>, Francesco De Francesco<sup>1,2\*</sup>, Gianfranco Nicoletti<sup>1</sup>,  
Francesca Paino<sup>2</sup>, Vincenzo Desiderio<sup>2</sup>, Virginia Tirino<sup>2</sup> and Francesco D'Andrea<sup>1</sup>

<sup>1</sup>Dipartimento di Scienze Ortopediche, Riabilitative, Traumatologiche e Plastico-Ricostruttive, Seconda Università degli Studi di Napoli, Italy;

<sup>2</sup>Dipartimento di Medicina Sperimentale, Sezione di Istologia ed Embriologia Medica, Tissue Engineering and Regenerative Laboratory (TERM), Seconda Università degli Studi di Napoli, Italy;

\*These authors contributed equally to this work

**Running title:** *In vivo* adipose and loose connective tissue formation

**Key Words:** human adipose stem cells, collagen scaffold, loose connective tissue, adipose tissue

### Acknowledgment

This study was supported by the PRIN 2008 to F.D.A.

The authors declare no potential conflicts of interest.

### Address for correspondence

Dott.ssa Virginia Tirino, PhD

Department of Experimental Medicine

Section of Histology and Embryology

Faculty of Medicine and Surgery

5 via L. Armani

80138 Naples (Italy)

Voice #+39 081 5666015

FAX # +39 081 5666014

e-mail: virginia.tirino@unina2.it

<sup>†</sup>This article has been accepted for publication and undergone full peer review but has not been through the copyediting, typesetting, pagination and proofreading process, which may lead to differences between this version and the Version of Record. Please cite this article as doi: [10.1002/jcb.24443]

Received 16 September 2012; Revised 21 October 2012; Accepted 24 October 2012

Journal of Cellular Biochemistry

© 2012 Wiley Periodicals, Inc.

DOI 10.1002/jcb.24443

## ABSTRACT

Stem cell based therapies for the repair and regeneration of various tissues are of great interest for a high number of diseases. Adult stem cells, instead, are more available, abundant and harvested with minimally invasive procedures. In particular, mesenchymal stem cells (MSCs) are multipotent progenitors, able to differentiate into bone, cartilage and adipose tissues. Human adult adipose tissue seems to be the most abundant source of MSCs and, due to its easy accessibility; it is able to give a considerable amount of stem cells. In this study, we selected mesenchymal stem cells co-expressing CD34 and CD90 from adipose tissue. This stem cell population displayed higher proliferative capacity than CD34<sup>-</sup>CD90<sup>-</sup> cells and was able to differentiate *in vitro* into adipocytes (PPAR $\gamma$ <sup>+</sup> and adiponectin<sup>+</sup>) and endothelial cells (CD31<sup>+</sup>VEGF<sup>+</sup>Flk1<sup>+</sup>). In addition, in methylcellulose without VEGF, it formed a vascular network.

The aim of this study was to investigate differentiation potential of human adipose CD34<sup>+</sup>/CD90<sup>+</sup> stem cells loaded onto commercial collagen sponges already used in clinical practice (Gingostat) both *in vitro* and *in vivo*.

The results of this study clearly demonstrate that human adult adipose and loose connective tissues can be obtained *in vivo*, highlighting that CD34<sup>+</sup>/CD90 ASCs are extremely useful for regenerative medicine.

## INTRODUCTION

Recently, the role of stem cells in tissue formation and regenerative medicine attracted considerable attention as a possible source of differentiated cells. In addition, the use of stem cells with biomaterial scaffolds and growth factors represents a suitable strategy for clinical therapies.

Stem cells are undifferentiated cells characterized by their ability to self-renew and to differentiate into multiple lineages (Zech *et al.*, 2011). In contrast to embryonic stem cells (ESCs), adult stem cells are immune-compatible and there are no ethical issues related to their use (Brignier and Gewirtz, 2010). Initially, adult stem cells from bone marrow (BM-MSCs) have been studied as an alternative source (Wang *et al.*, 2011; Conget and Minguell, 1999). These cells can differentiate into adipocytes, chondrocytes, osteoblasts, myoblasts and neurons, among others (Schilling *et al.*, 2007; Ferrari *et al.*, 1998; Woodbury *et al.*, 2000). However, the clinical use of BM-MSCs presented several problems, including pain, morbidity, and low cell number upon harvest. Therefore, many researchers investigated alternative sources of MSCs in different tissues such as dental pulp (Graziano *et al.*, 2008), Wharton jelly (Romanov *et al.*, 2003), amniotic fluid (Roubelakis *et al.*, 2011) and adipose tissue (Zuk *et al.*, 2002).

Adipose tissue derived from mesenchyme represents an optimal source of adult stem cells, sharing many characteristics with bone marrow, including extensive proliferation and ability to undergo multi-lineage differentiation (Witkowska-Zimmy and Walenko, 2011). It contains numerous cells types, including adipocytes, preadipocytes, and vascular endothelial cells. In our previous experiments, using human liposuction or lipectomy, we have identified a stem cell population that expressed MSCs markers, including CD34, CD90, CD29, CD44, and CD105, but they do not express the hematopoietic marker CD45. Cloning studies have shown that this cells have multi-lineage differentiation potential (Zuk *et al.*, 2001; Halvorsen *et al.*, 2001; Erickson *et al.*, 2002). Moreover, we have demonstrated that CD34<sup>+</sup>/CD90<sup>+</sup> stem cell population is capable of: i) differentiating into endothelial cells and forming capillary-like structures in a semisolid medium with or without the use of angiogenic factors; ii) forming sphere clusters in a serum-free medium

supplemented with bFGF and EGF; iii) showing higher telomerase activity than differentiated cells and iv) displaying a side population phenotype (De Francesco *et al.*, 2009). The combination of these properties, and the large quantity of cells that can be obtained from fat, suggest that this tissue will be a useful tool in biotechnology and regenerative medicine.

Currently, plastic surgeon uses autologous tissue grafts, including dermal and fat grafting, free and pedicle flap or synthetic implants for the reconstruction and/or augmentation of soft tissue (Ferraro *et al.*, 2011; Rosson *et al.*, 2010). Various levels of clinical success have been reported with the use of autologous soft tissue. However, donor-site morbidity remains the principal liability. In addition, autologous tissue grafts have a tendency to lose volume with an average of 40 to 60 % reduction in tissue volume gradually. Engineered soft tissue from stem cells has the potential to overtake most of lack associated with autologous soft-tissue grafts (Stosich and Mao, 2005). Biodegradable scaffolds should ideally provide the initial required blood flow, and then gradually degrade to leave a graft composed only of new tissue. Researchers have developed several studies of adipose tissue engineering by using BM-MSCs and ASCs. Various scaffold materials have been analyzed both *in vitro* and *in vivo*, using synthetic or natural prefabricated scaffolds such as poly(lactic-co-glycolic acid) (Morgan *et al.*, 2009), poly(ethylene glycol)-based hydrogel (Stosich and Mao, 2005), silk fibroin scaffold (Mauney *et al.*, 2007), hyaluronic acid (Hemmerich *et al.*, 2008) and collagen (von Heimburg *et al.*, 2001; von Heimburg *et al.*, 2003), but, up to now, obtained data are still not suitable for clinical application.

The ideal scaffold must have certain characteristics: i) easy to shape it in the tissue defect; ii) good capacity of growing and maturing stem cells; and iii) reabsorption time compatible with the time required for tissue formation.

Type I collagen, the main fibre of the extra-cellular matrix in connective tissues, has shown a high affinity for the living organism and has also been extensively used as scaffold for adipose tissue engineering (Rubin *et al.*, 2007; Itoi *et al.*, 2010).

Therefore, the aim of this study was to evaluate the distribution *in vitro* of cultured CD34<sup>+</sup>CD90<sup>+</sup> ASCs into commercial solid collagen sponges, and study the differentiation potential *in vivo* of the grafted ASCs loaded on these scaffolds.

## MATERIALS AND METHODS

### *In vitro tissue culture*

Subcutaneous adipose tissue from abdomen and mammary region was obtained with informed consent, approved by our Internal Ethical Committee (Second University Ethical Committee) from 52 female donors aged 18 to 43 years (mean years 30,5) that had endured elective procedures for plastic surgery. Adipose tissue was obtained by lipectomy or liposuction in the Plastic and Reconstructive Surgery Clinic of the Second University of Naples. The adipose tissue was placed in a physiological solution (0.9% NaCl), washed twice in PBS (phosphate saline buffer: 137mM NaCl, 2.7mM KCl, 10mM Na<sub>2</sub>HPO<sub>4</sub>, 1.8mM KH<sub>2</sub>PO<sub>4</sub>), scraped, and placed in a digestion solution: collagenase type I (3mg/ml) and dispase (4mg/ml) in PBS at 37°C in agitation for 60 min. The digestive solution was filtered through 70 µm filters (Becton & Dickinson, Sunnyvale, CA).

After filtration and washing, the pellet was re-suspended in erythrocyte lysis buffer (155mM NH<sub>4</sub>Cl, 10mM KHCO<sub>3</sub>, 0.1mM EDTA, pH 7.3) for 10 min at room temperature. The cell suspension was centrifuged at 1300 rpm for 7 min and the pellet re-suspended in 5ml Dulbecco's modified Eagle's medium (DMEM) with 10% fetal bovine serum (FBS), 2mM L-glutamine, 100U/ml penicillin, and 100µg/ml streptomycin and seeded in 25cm<sup>2</sup> flasks. Flasks were incubated at 37 °C under 5% CO<sub>2</sub> and the medium changed twice a week. Cells reached confluence in 5-7 days. Experiments were performed in quadruplicate.

Cells were detached using trypsin-EDTA (200mg/L EDTA, 500mg/L trypsin; Cambrex). At least 200,000 cells were incubated with primary antibody for 30 min at 4°C, washed twice in PBS, and incubated with a secondary antibody. Alternatively, cells were incubated directly with fluorescent-conjugated antibodies for 30min at 4°C, washed, and re-suspended in 0.6ml PBS. Samples were

analyzed at day 0 (day of surgery), day 7, day 15, and day 30 by flow cytometry using a FACS Aria II (Becton & Dickinson, Mountain View, CA, USA). The antibodies used in this study were: anti-CD117 PE (c-kit) (Miltenyi-Biotech, Calderara di Reno, Bologna, Italy); anti-CD34 FITC and PE (Miltenyi-Biotech); anti-CD90 FITC (BD Pharmingen, Buccinasco, Milano, Italy); anti-CD105 FITC (Santa Cruz, CA, USA); anti-CD29 PE (Miltenyi-Biotech); anti-CD31 FITC (Miltenyi-Biotech); anti-CD133 PE (Miltenyi-Biotech); anti-hVEGF (Santa Cruz, CA); anti-VEGFR-2 (Santa Cruz); anti-CD54 PE (Miltenyi-Biotech); anti-CD44 FITC (Miltenyi-Biotech), anti-CD45 Cy and PE (BD Pharmingen, Buccinasco, Milano, Italy); and anti-CD14 PE (Miltenyi-Biotech). CD34<sup>+</sup>/CD90<sup>+</sup> cells were sorted for experiments. The purity of sorted populations was routinely of 90%.

#### *Growth Curve*

CD34<sup>+</sup>CD90<sup>+</sup>, CD34<sup>-</sup>CD90<sup>-</sup> and unsorted cells were plated at a density of  $8,0 \times 10^4$  cells/well in 6-well plates. Every twelve hours, cells were harvested and re-suspended in PBS for 7 days. An aliquot of cell suspension was counted under a microscope at 20 X magnification. The number of viable cells for each experimental condition was counted and represented on a linear graph. The doubling time (DT) was determined from the growth curves or by using the formula:

$$DT = (t - t_0) \log 2 / (\log N - \log N_0)$$

where  $t$  and  $t_0$  were the times at which the cells were counted, and  $N$  and  $N_0$  were the cell numbers at times  $t$  and  $t_0$ , respectively.

#### *In vitro adipogenic differentiation of ASCs*

Cells were induced in the following adipogenic medium for 2-3 weeks: DMEM supplemented with 10% FBS plus dexamethasone (1  $\mu$ M; Sigma), human recombinant insulin (10  $\mu$ M; Sigma), indomethacin (200  $\mu$ M; Fluka,) and 3-isobutyl-1-methyl-xantine (IBMX) (0.5 mM; Sigma). Cells cultured in basal medium (see above) were used as controls.

### *In vitro angiogenic differentiation of ASCs*

To analyze *in vitro* capillary-like morphology,  $2 \times 10^5$  -  $5 \times 10^5$  cells/ml were plated in 24-well plates in a semisolid growth medium that consisted of 0.9% methylcellulose in DMEM, 30% FBS, 1% bovine serum albumin (BSA),  $10^{-4}$ mol/L mercaptoethanol, and 2 mmol/L L-glutamine. In parallel experiments, cultures were stimulated in addition with Vascular Endothelial Growth Factor (VEGF, 50 ng/ml). All cultures were performed in triplicate, incubated at 37° C under 5% CO<sub>2</sub> and left for 7 days to develop a capillary-like morphology.

### *Preparation of collagen sponges for in vitro analyses and in vivo transplantation*

After trypsinisation with 0.05% trypsin-EDTA, either unsorted ASCs or ASCs CD34<sup>+</sup>/CD90<sup>+</sup>, at a density of  $2 \times 10^5$  cells/injection were seeded onto Collagen sponge of 10x10 mm size and cultured in DMEM at 10% FBS. Gingistat (GabaVebas, San Giuliano Milanese, Italy) used in this study, is a commercial type 1 collagen, made of native lyophilized collagen from animal origin (equine) with interconnected pores with an average size of 300µm. Collagen sponges were cut in 10x10mm squared pieces and then sterilized under UV exposure. These constructs were incubated for 48h at 37 °C to allow the cells to attach to the scaffolds. For *in vitro* examinations of cell adherence and proliferation on carriers, sponges were analysed after 7 and 14 days, and histologically evaluated. Cell viability was routinely checked by Calcein AM at the concentration of 250µM in culture medium for 30 min at 37°C. This dye passes through the cell membrane of viable cells. After calcein permeates into the cytoplasm, it is hydrolyzed by esterases to calcein, which remains inside the cell. The excitation and emission wavelengths of calcein were of 490 nm and 515 nm, respectively. For *in vivo* studies, the construct was left undisturbed in DMEM in the incubator for 48h until implantation.

### *In vivo experimental model for grafting*

For *in vivo* experiments, we have used ASCs of subcutaneous adipose tissue from abdomen and breast of 2 healthy female donors (mean years 30) with a normal range of BMI. We have

previously obtained informed consent, approved by our Internal Ethical Committee (Second University Ethical Committee).

Twenty-four hours after seeding of the cells to the scaffold, unsorted ASCs and CD34<sup>+</sup>/CD90<sup>+</sup> ASCs (at a density of  $2 \times 10^5$  cells/injection)/sponge matrix constructs were engrafted subcutaneously to the right and left scapular area of 4-week-old immune-compromised athymic nude mice (NU/NU nude mice, Charles River, Ballina, Italy) according to the scheme reported in Table 1. Mice were operated under aseptic conditions and inhalation anesthesia. In each of these animals, the cell-loaded sample was transplanted to the right and the control scaffold to the left scapular area through separate incisions. After 4 weeks, the animals were killed by an overdose of gaseous anesthetic and the graft was explanted. The experimental protocols have been evaluated and approved by our Internal Animal Ethic Committee (Second University Animal Ethic Committee). Explanted tissue volume was calculated by the formula  $(l \times w^2)/2$ . The injection experiments were made in triplicates.

#### *Haematoxylin & Eosin and Mallory's Trichromic Staining*

Constructs derived from implantations in mice were fixed with 4% paraformaldehyde for 24 hours at 4°C, washed in PBS, incubated in 30% sucrose for 3 days, washed in PBS and then embedded in an optimal cutting temperature (OCT-purchased from Bio-Optica, Milan, Italy). Finally, they were stored at -80°C for Mallory's trichromic and haematoxylin and eosin (H&E) staining. The frozen sections in the OCT were cut into 5µm thick slices, washed in distiller water for 10 min. For haematoxylin and eosin staining, sections were placed in haematoxylin for 5 minutes. After three washes by distilled water, samples were placed in spring water for 20 min. Then, the samples were placed in eosin acidified with acetic acid for 30 seconds. Finally, they were placed in alcohol 95°, 75° and 95° and mounted with DPX. For Mallory's trichromic stain, the sections were stained in acid fuchsine for 1 min, methylene blue-orange G and oxalic acid solution for 25 min, and phosphotungstic acid for 15 min; fibrils of collagen are blue, nuclei red, and cytoplasm is orange. Finally, they were placed in alcohols 95°, 75° and 95° and mounted with DPX.

### *Immunofluorescence and Immunohistochemistry*

Frozen sections in OCT were cut into 5  $\mu\text{m}$  thick slices, washed in distiller water for 10 min. For immunofluorescence, tissue sections were incubated in PBS at 5% milk for 60 min at room temperature. After a double washing in PBS for 10 min at room temperature, cells were incubated overnight at 4°C with monoclonal anti-human antibodies (diluted 1:100 in PBS). Sections were washed in PBS three times for 10 min at room temperature and incubated for 90 min at 4°C with the secondary FITC- or PE-conjugated antibody (diluted 1:200 in PBS) (AbCam, Cambridge, UK). Moreover, sections were stained with DAPI (4', 6-diamidino-2-phenylindole) (Invitrogen, San Giuliano Milanese, Milan, Italy) diluted 1:10000 (5 $\mu\text{g}/\text{ml}$ ) in PBS for 7 min at room temperature. Tissue sections incubated for 90 min at 4°C only with conjugated secondary antibodies were used as negative control. The sections were then observed under a fluorescence microscope (Nikon Instruments Italia, Calenzano, Firenze, Italy). Immunohistochemical analyses were performed with a DAKO CYTOMATION kit (En Vision+System-HRP-AEC, Dako Italia, Milan, Italy). Antibodies used were: I Class HLA (AbCam), anti-adiponectin (AbCam), Glut4 (AbCam), PPAR $\gamma$  (AbCam), Collagen Type I (AbCam) and VEGF (AbCam).

### *RNA isolation and polymerase chain reaction*

Frozen tissues were homogenized in TRIzol® lysis buffer using a handheld homogenizer for at least 45 seconds. Then, the samples were centrifuged at 12,000 x g for 10 minutes at 4°C. The supernatant was processed for the phase separation in according with the manufacturer's instructions. RNA concentration and purity were determined by A260 and A260/A280 ratios, respectively. The integrity of total RNA was assessed on standard 1% agarose/formaldehyde gels. The RNA samples were treated with DNase I to remove residual traces of DNA. cDNA was obtained from 1 $\mu\text{g}$  of total RNA, using reverse transcriptase (Promega Italia srl, Milan, Italy) and random primers (Promega) in a final volume of 20 $\mu\text{l}$ . cDNAs (1 $\mu\text{l}$  for each sample) were amplified by PCR using the primer sequences as follows:

*GADPH*: fw 5'-AGCCGCATCTTCTTTTGCCTC-3', rw 5'-TCATATTTGGCAGGTTTTTCT-3';

*human adiponectin*: fw 5'-CAACATTCCTGGGCTGACT-3', rw 5'-CCTGTGAAGGTGGAGTCATT-3';

*human PPAR-γ*: fw 5'-ACAGCAAACCCCTATTCCATGC-3', rw: 5'-ATTACGGAGAGATCCACGGAGC-3';

*human VEGF*: fw 5'-TGACAGGGAAGAGGAGGAGA-3', rw 5'-CGTCTGACCTGGGGTAGAGA-3';

*human fibronectin*: fw 5'-CTGGGATGCTCCTGCTGTCAC-3', rw 5'-CTGTTTGATCTGGACCTGCAG-3';

*human collagen type I*: fw 5'-TACCATGACCGAGACGTGTG-3', rw 5'-ATAAGACAGCTGGGGAGCAA-3'.

The RT-PCR products were separated by 2% agarose gel electrophoresis, stained with ethidium bromide, and photographed under UV illumination.

#### *Statistical analysis*

Student t-test (two tailed) was used for statistical evaluation. Level of significance was set at  $p < 0.05$ .

## **RESULTS**

### *In vitro experiments*

#### *In vitro characterization of ASCs*

Adipose tissue was obtained by lipectomy or liposuction. After enzymatic digestion, samples of 1,000,000 cells were collected from cell suspension and cultured. After three days of culture, adherent cells acquired a fibroblast-like or polygonal shape morphology. Cells were analysed by flow-cytometry at different times. At day 0, as previously demonstrated by us (De Francesco *et al.*, 2009), we detected stromal cells co-expressing CD117 (~5%) and CD34 (~7%), mesenchymal stem cells, co-expressing CD29 (β1-integrin) (~72%), CD34 (~25%), CD90 (Thy-1)

(~30%), and CD105 (endoglin) (~5%) and endothelial stem cells, co-expressing CD31 (~6%), CD133 (~8%), CD44<sup>+</sup>CD54<sup>+</sup> (~5%) and Flk-1 (VEGFR-2) (~45%) (Fig. 1A,B). Only CD34<sup>+</sup>CD90<sup>+</sup> cells (~25%) were sorted (Fig. 1C) and cultured for both *in vitro* and *in vivo* experiments. Co-expression of CD34 and CD90 markers was ~53% and ~80%, at 7 and 15 days (Fig. 1D), respectively while after 30 days, their expression was ~8%. CD90 expression was 80% (Fig. 1E). CD34<sup>+</sup>CD90<sup>+</sup> cells exhibited: i. a higher proliferative capacity than CD34<sup>+</sup>CD90<sup>+</sup> cells with a mean doubling time of 72 hours with respect to 86 and 100 hours of unsorted cells and CD34<sup>-</sup>CD90<sup>-</sup> cells, respectively (Fig. 2); ii. In adipogenic medium, some unsorted cells acquired the morphology of fibroblasts, whereas other acquired the morphology of multi-vacuolar adipocytes (Fig. 3A), positive for adiponectin, (Fig. 3B). On the contrary, CD34<sup>+</sup>CD90<sup>+</sup> cells acquired the typical morphology of adipocytes (Fig. 3C) positive, for adiponectin and PPAR- $\gamma$  already after 24h of incubation at the immunohistochemistry (Fig. 3D,E); iii. In methylcellulose without VEGF, they formed a vascular network (Fig. 3F) while in standard medium, they acquired an endothelial morphology and expressed high levels of endothelial markers such as CD90 (~97%), CD44 (~90%), CD54 (~90%), VEGF (~70%), CD133 (~18%), and Flk-1 (~60%) (Fig. 3G).

#### *Cell attachment to the scaffold and in vitro observations*

Each scaffold was loaded with  $2 \times 10^5$  sorted ASCs (CD34<sup>+</sup>CD90<sup>+</sup>), placed in the incubator for 48 h and analyzed afterwards for cell attachment. The range of unattached CD34<sup>+</sup>CD90<sup>+</sup> cells was approximately 5% in all sponges, as well as for unsorted cells. Adherence and viability of CD34<sup>+</sup>CD90<sup>+</sup> ASCs to sponges was high, as shown by Calcein staining (Fig. 4A,B). In order to evaluate cell proliferation, constructs were analyzed by light-inverted microscopy (Fig. 4C,D) and by histological staining after 7 and 14 days (Fig. 4E,F). Light-inverted microscopy and H&E staining on serial sections of ASCs seeded onto the Gingistat collagen sponge demonstrated that the cells were homogeneously distributed throughout the whole sponge. Moreover, at day 14, a strong spreading and the proliferation of ASCs was observed with respect to day 7.

## ***In vivo experiments***

### *In vivo adipose tissue and loose connective formation*

Unsorted ASCs or ASCs CD34<sup>+</sup>/CD90<sup>+</sup>, at a density of 2x10<sup>5</sup> cells/injection were seeded onto Collagen sponge of 10x10 mm size (Fig. 5A). After 30 days of implantation, we extracted the constructs sorted ASCs/collagen from athymic nude mice (Fig. 5B,C) (mean tissue volume 125,94 ± 1 mm<sup>3</sup>), unsorted ASCs/collagen (mean tissue volume 119.52 ± 0.8 mm<sup>3</sup>) (Table 2). Collagen without cells used as controls, was not detectable on the dorsum of mice because it has been completely reabsorbed (Fig. 5D) as well as unsorted and sorted ASCs. The grafts with unsorted ASCs showed formation of adipose tissue appearing almost avascular (Fig. 6A). All collagen sponges with ASCs CD34<sup>+</sup>CD90<sup>+</sup> showed a thin layer of macroscopically yellow tissue and new vessels on the top. This layer was tightly connected to the sponge.

H&E staining showed that, within the sorted ASCs/collagen constructs, stem cells differentiated towards adipocytes (fig. 6B). Neo-formed adipose tissue was made of fat cells arranged into groups or lobules separated by collagenic septa. Moreover, each individual fat cell was surrounded by a network of loose connective tissue (Fig. 6C). The formation of this connective tissue surrounding the adipose tissue was observed in all histological serial slides. The fibres interlaced themselves loosely to form a network with interstices or spaces between them. The tissue was characterized by cellular, fibrous and amorphous components. Collagenic fibres predominated and constituted the bulk of the loose connective tissue as shown by Mallory's tri-chromic staining (Fig.6D). Amorphous material was transparent and homogenous under the optical microscope and occupied numerous small areas (areolas) in which no structures were orderly seen.

Class I HLA positive staining (Fig. 6E) clearly demonstrated the human origin of the adipose tissue obtained excluding any murine contamination. In addition, the tissue showed a strong positivity for adiponectin (Fig. 6F), Glut-4 (Fig. 6G), PPAR- $\gamma$  (Fig. 6H), Collagen Type I (Fig. 6I) that are specific markers of both adipose and loose connective tissues. Moreover, the positivity of some areas for the endothelial markers such as VEGF, demonstrated that the tissue was well

vascularised: in fact, a rich network of blood capillaries in and among the lobules was evidenced (Fig. 6J).

#### *Expression of adipogenic and loose connective differentiation-associated genes*

RT-PCR analyses for adipose differentiation-associated genes in sorted ASCs grown on Collagen sponge, after 30 days from implantation, revealed that PPAR- $\gamma$  and adiponectin expressions were detectable. Moreover, VEGF levels were highly expressed in all the samples, confirming the neo-angiogenesis of the neo-formed adipose tissue. In addition, expression of mRNA for fibronectin and collagen type I showed the generation of loose connective tissue associated to neo-formed adipose tissue (Fig. 7).

## **DISCUSSION**

Regenerative medicine and tissue engineering both harness the potency of human cells to repair, regenerate, and even recreate tissue and organs with the goal of restoring their architecture and functionality. Soft-tissue loss can be caused by trauma, tumour resection, extensive deep burns, as well as congenital or acquired anomalies and it presents an ongoing challenge in plastic and reconstructive surgery. Standard approaches to soft-tissue reconstruction include autologous tissue flaps, autologous fat transplantation and alloplastic implants (Bucky *et al.*, 2008; Megerle *et al.*, 2011). All of these approaches have disadvantages, including donor-site morbidity, implant migration and absorption and foreign body reaction. ASCs are currently used for the engineering of substitutes that will ultimately be utilized in several fields of reconstructive surgery.

The availability of progenitors within the adipose stromal fraction (CD34<sup>+</sup>/CD90<sup>+</sup> stem cells) for building neo-tissue types and the understanding of vectors that govern their differentiation emerge as important factors for successful neo-tissue construction. To successfully create a 3D structure capable of supporting tissue formation, several key roles of tissue engineering need to be considered, including pores with interconnected architecture; material degradability; biocompatibility with the seeded cells to enhance cellular activity (O'Brien *et al.*, 2005; Kuberka *et*

*al.*, 2002). Therefore, the formation of a new tissue in the 3D matrix structure is highly influenced by the chemical composition of the scaffold (Cassel *et al.*, 2001; Liu *et al.*, 2011). The extracellular matrix (ECM) is recognized as a highly hydrated network that comprises several main effectors. Its architecture provides not only an instructive cue for the conversion and spatial orientation of precursor cells (CD34<sup>+</sup>/CD90<sup>+</sup> stem cells) into a functional tissue but also functional properties including osmosis, molecular and nutrient transport. Collagen, being a major protein of the natural extracellular matrix (ECM), has been commonly used in the form of a gel for tissue engineering in spite of limitations principally related to cell-mediated interactions (Natesan *et al.*, 2010). It is a physiological component, with a fast biodegrading rate and a low mechanical strength. In fact, many surgeons often add other components to collagen in order to improve its stability (Labbè *et al.*, 2011).

Multiple independent groups have examined the surface immunophenotype of ASCs isolated from human (Yoshimura *et al.*, 2006; Gronthos *et al.*, 2001; Mitchell *et al.*, 2006). Our previous studies have evidenced that adipose tissue houses CD34<sup>+</sup>CD90<sup>+</sup> multipotent stem cells (D'Andrea *et al.*, 2008; De Francesco *et al.*, 2009). No significant difference between subcutaneous and omental adipose tissue is demonstrated concerning the expression of these markers. There is some controversy in the literature regarding expression of CD34 by ASCs. CD34 is used as marker of hematopoietic stem and vascular endothelial cells. Our data suggest that expression of this molecule is present in cultured ASCs (De Francesco *et al.*, 2009) but, by contrast, Zuk *et al.* (2002) and De Ugarte *et al.* (2003), have both reported low-to-absent levels of CD34 expression. In our studies, CD34<sup>+</sup>/CD90<sup>+</sup> stem cells, within human adipose tissue, are numerous. *In vitro*, these cells are able to proliferate and differentiate into mesodermal (such as osteogenic, chondrogenic, adipogenic and myogenic) and non mesodermal (such as endothelial cells, epithelial cells) lineages, under appropriate stimuli. Moreover, this cell population is capable of differentiating into endothelial cells with or without the use of angiogenic factors, then CD34<sup>+</sup>/CD90<sup>+</sup> stem cells are an endothelial commitment (Miranville *et al.*, 2004; De Francesco *et al.*, 2009).

On the basis of our *in vitro* studies of ASCs, tissue regeneration and differentiation experiments using CD34<sup>+</sup>/CD90<sup>+</sup> stem cells with appropriate scaffold have been performed *in vivo*. In this study, we describe the *in vivo* formation of both loose connective and adipose tissues, starting from ASCs co-expressing CD34 and CD90 markers and seeded on a biocompatible commercial scaffold of type I collagen (Gingostat). This lyophilized collagen sponge is widely used as a scaffold for spontaneous bone regeneration, mainly in periodontal diseases. Therefore, its wide use reflects the good histo-compatibility of this material with cells. The result of our study suggest that ASCs/collagen sponge constructs better support *in vivo* engraftment, as indicated by vascularization after *in vivo* transplantation. In addition, ASCs/collagen sponge better support formation of both loose connective and adipose tissues, as indicated by higher expression levels of specific markers.

Studies by Von Heimburg (2001) have demonstrated that the transplantation of isolated and cultured preadipocytes seeded on freeze-dried collagen scaffolds resulted in well-vascularised adipose-like tissue. Here, we used CD34<sup>+</sup>/CD90<sup>+</sup> ASCs demonstrating that these stem cells are easily absorbed and distributed among the whole collagen scaffold. The structural characteristics and architecture of the collagen sponge with interconnected pores sustained and enhanced both cell proliferation and spreading without lose so facility to differentiate. Biodegradable scaffold used in the experiments was designed to meet tissue engineering imitating the *in vivo* three-dimensional extracellular architecture and to provide to precursors the signals for develop in desirable phenotypes. In the xenograft experiments, analyses of the new-formed tissue through histological staining revealed that the collagen, on which CD34<sup>+</sup>/CD90<sup>+</sup> ASCs were loaded, induced the adipose differentiation with complete absorption of scaffold and loose connective tissue, suggesting CD34<sup>+</sup>/CD90<sup>+</sup> ASCs have high affinity for the Type I collagen sponge. In addition, we found an extensive formation of new, tightly integrated vessels throughout the neoformed tissue. By the contrast, in xenografts in which non-sorted ASCs were used, only adipose tissue was obtained.

Accepted Article

Investigators have postulated a number of mechanisms through which MSCs can be used to repair and regenerate tissues. We may hypothesize that CD34<sup>+</sup>CD90<sup>+</sup> ASCs may secrete cytokines and growth factors that stimulate recovery in a paracrine manner. They could modulate the “stem cell niche” of the host by stimulating the recruitment of endogenous stem cells to the site and promoting their differentiation along the required lineage pathway (Gimble *et al.*, 2007). In a related manner, they might provide antioxidants chemicals, free radical scavengers, and chaperone/heat shock proteins at an ischemic site. As a result, toxic substances released into the local environment would be removed, thereby promoting recovery of the surviving cells. Exciting studies have suggested that transplanted bone marrow–derived MSCs can deliver new mitochondria to damaged cells, thereby rescuing aerobic metabolism (Spees *et al.*, 2006). It may develop that similar studies in ASCs will uncover a comparable ability to contribute mitochondria. Therefore many and different mechanisms highlight the great potentiality of CD34<sup>+</sup>CD90<sup>+</sup> ASCs versus unsorted ASCs in transplantations.

Shape and dimensions are of critical importance to plastic surgery applications of engineered soft tissue. Creation and maintenance of the shape and dimensions of the engineered soft tissue are perhaps best performance by scaffold biomaterials. Collagen scaffold is biodegradable and fully absorbable when transplanted subcutaneously. Moreover, for effective MSC seeding and proliferation needed to generate new tissue, the scaffold must be porous and three-dimensional. The structure of collagen sponge used in this study confirmed their porous 3D architecture. Pores of the collagen matrix have an average size of 300µm. In this study, analysis of CD34<sup>+</sup>/CD90<sup>+</sup> cells by light-inverted microscopy confirmed their satisfactory seeding, proliferation and differentiation within pores.

This is the first time that adipose and loose connective tissues have been obtained, and that a complete physiological and functional reconstruction of tissues has been generated reinforcing the hypothesis that CD34<sup>+</sup>/CD90<sup>+</sup> ASCs could be used in regenerative medicine. Moreover, the use of the collagen as scaffold with CD34<sup>+</sup>/CD90<sup>+</sup> ASCs is advantageous for different motivations i. commercial available; ii. easily obtainable; iii. avoid donor morbidity; and iv. easily formed into the

desired shape and volume. For the repair of tissue defects, especially after cancer ablation, typically it is essential to restore both the volume and shape. Therefore, to correct such defects is crucial to recreate a three-dimensional multilayer especially for the tissue shape. In an in vivo study of engineering adipose tissue using MSC from human bone marrow, Stosich and Mao (2011) have used a poly(ethylene glycol) scaffold to maintain a stable shape; instead Lin *et al.* (2008) have used non-biodegradable polyglycolic acid scaffold to keep the predefined shape. Other researchers have used natural scaffolds such as fibrin (Mauney *et al.*, 2007) and gelatin sponges (Kimura *et al.*, 2003). The main drawback of these scaffolds is the fast resorption rate that does not allow the achievement of an adequate volume so as not allow the organization of an appropriate shape.

The novelty of this research is represented by the formation not only of adipose tissue starting from ASCs, differently from Von Heimburg (2001) and co-workers who used preadipocytes, but also by the formation of loose connective tissue, as shown in this study using E&E and Mallory's trichromic stainings, RT-PCR and immunofluorescence analyses for adiponectin, PPAR- $\gamma$ , Glut-4, fibronectin, and collagen I. In particular, we have reconstructed both connective and adipose tissues from cultured human ASCs differentiating into adipocytes, endotheliocytes, and connective cells, able to secrete cellular matrix components, including collagen fibres, fibronectin and amorphous substance.

In our study, we used a biodegradable and bio-absorbable collagen matrix, so that both the volume and the shape of the defect can be reconstructed using a predefined scaffold at the outset. The slow biodegradation of the scaffold allows maintenance of the scaffold volume of adipose tissue newly generated by SVF.

On the basis of both in vitro and in vivo experiments and on the basis of preclinical studies available in the literature (Mizuno, 2009), ASCs/collagen construct could be applied to various clinical fields, such as treatment of chronic ulcers, cancer ablation, post-traumatic calvarial bone defect, but also could be an alternative to soft tissue augmentation surgery including cosmetic breast augmentation.

In conclusion, with this study we provided evidence that ASCs differentiate *in vivo* into both adipose and loose connective tissues, when loaded on collagen scaffold. This demonstrates the feasibility of collagen scaffold with sorted ASCs (CD34<sup>+</sup>/CD90<sup>+</sup>) as transplantation vehicle for adipose and loose connective tissue engineering and its employment for reconstruction in plastic and reconstructive surgery. Collagen scaffolds were demonstrated to be suitable materials for soft-tissue regeneration; they maintained volume when seeded with sorted ASCs. The material had a progressive rate of biodegradation, lacked cytotoxicity and did not induce a systemic immune response or chronic inflammation in this human *in vivo* model. The observed tissue integration is promising and in addition of this the angiogenic formation suggest that collagen scaffold would be an efficient biomaterial for adipose and loose connective growth and function.

## References

1. Brignier AC and Gewirtz AM. 2010. Embryonic and adult stem cell therapy. *J Allergy Clin Immunol* 125: S336-44.
2. Bucky LP and Percec I. 2008. The science of autologous fat grafting: views on current and future approaches to neoadipogenesis. *Aesthet Surg J* 28:313-21.
3. Cassell OC, Morrison WA, Messina A, Penington AJ, Thompson EW, Stevens GW, Perera JM, Kleinman HK, Hurley JV, Romeo R, Knight KR. 2001. The influence of extracellular matrix on the generation of vascularized, engineered, transplantable tissue. *Ann NY Acad Sci*, 944:429-42.
4. Conget PA and Minguell JJ. 1999. Phenotypical and functional properties of human bone marrow mesenchymal progenitor cells. *J Cell Physiol* 181:67-73.
5. D'Andrea F, De Francesco F, Ferraro GA, Desiderio V, Tirino V, De Rosa A, Papaccio G. 2008. Large-scale production of human adipose tissue from stem cells: a new tool for regenerative medicine and tissue banking. *Tissue Eng Part C Methods* 14:233-42.
6. De Francesco F, Tirino V, Desiderio V, Ferraro G, D'Andrea F, Giuliano M, Libondi G, Pirozzi G, De Rosa A, Papaccio G. 2009. Human CD34/CD90 ASCs are capable of growing as sphere clusters, producing high levels of VEGF and forming capillaries. *PLoS One*, 4:e6537.
7. De Ugarte DA, Morizono K, Elbarby A, Alfonso Z, Zuk PA, Zhu M, dragoo JL, Ashjian P, Thomas B, Benhaim P, Chen I, Fraser J, Hedrick MH. 2003. Comparison of multi-lineage cells from human adipose tissue and bone marrow. *Cells Tissues Organs* 174:101-109.
8. Erickson GR, Gimble JM, Franklin DM, Rice HE, Awad H, Guilak F. 2002. Chondrogenic potential of adipose tissue-derived stromal cells in vitro and in vivo. *Biochem Biophys Res Commun* 290:763–769.
9. Ferrari G, Cusella-De Angelis G, Coletta M, Paolucci E, Stornaiuolo A, Cossu G, Mavilio F. 1998. Muscle regeneration by bone marrow-derived myogenic progenitors. *Science* 279: 1528-30. Erratum in: *Science* 281:923.
10. Ferraro GA, De Francesco F, Tirino V, Cataldo C, Rossano F, Nicoletti G, D'Andrea F. 2011. Effects of new centrifugation method on adipose cell viability for autologous fat grafting. *Aesthetic Plast Surg* 35: 341-8.
11. Gimble JM, Katz AJ, Bunnell BA. 2007. Adipose-Derived Stem Cells for Regenerative Medicine. *Circ Res* 100:1249-60.

12. Graziano A, d'Aquino R, Laino G, Papaccio G. 2008. Dental pulp stem cells: a promising tool for bone regeneration. *Stem Cell Rev* 4:21-6. Erratum in: *Stem Cell Rev* 4:65.
13. Gronthos S, Franklin DM, Leddy HA, Robey PG, Storms RW, Gimble JM. 2001. Surface protein characterization of human adipose tissue-derived stromal cells. *J Cell Physiol* 189:54-63.
14. Halvorsen YD, Franklin D, Bond AL, Hitt DC, Auchter C, Boskey AL, Paschalis EP, Wilkison WO, Gimble JM. 2001. Extracellular matrix mineralization and osteoblast gene expression by human adipose tissue-derived stromal cells. *Tissue Eng* 7: 729–741.
15. Hemmrich K, Van de Sijpe K, Rhodes NP, Hunt JA, Di Bartolo C, Pallua N, Blondeel P, von Heimburg D. 2008. Autologous in vivo adipose tissue engineering in hyaluronan-based gels - a pilot study. *J Surg Res* 144:82-88.
16. Itoi Y, Takatori M, Hyakusoku H, Mizuno H. 2010. Comparison of readily available scaffolds for adipose tissue engineering using adipose-derived stem cells. *J Plast Reconstr Aesthet Surg* 63:858-64.
17. Kimura Y, Ozeki M, Inamoto T, Tabata Y. 2003. Adipose tissue engineering based on human preadipocytes combined with gelatin microsphere containing basic fibroblast growth factor. *Biomaterials*. 24:2513-2521.
18. Kuberka M, von Heimburg D, Schoof H, Heschel I, Rau G. 2002. Magnification of the pore size in biodegradable collagen sponges. *Int J Artif Organs* 25:67-73.
19. Labbè B, Marceau Fortier G, Fradette J. 2011. Cell sheet technology for tissue engineering: The self-assembly approach using adipose-derived stromal cells. *Methods Mol Biol* 702:429-41.
20. Lin SD, Wang KH, Kao AP. 2008. Engineered adipose tissue of predefined shape and dimensions from human adipose-derived mesenchymal stem cells. *Tissue Eng Part A* 14:571-581.
21. Liu X, He J, Zhang S, Wang XM, Liu HY, Cui FZ. 2011. Adipose stem cells controlled by surface chemistry. *J Tissue Eng Regen Med* doi: 10.1002/term.498.
22. Megerle K and Sauerbier M. 2011. Reconstructive treatment of soft tissue sarcoma of the upper extremity. *J Hand Surg Am* 36:1241-7.
23. Mauney JR, Nguyen T, Gillen K, Kirker-Head C, Gimble JM, Kaplan DL. 2007. Engineering adipose-like tissue *in vitro* and *in vivo* utilizing human bone marrow and adipose-derived mesenchymal stem cells with silk fibroin 3D scaffolds. *Biomaterials* 28:5280-5290

24. Miranville A, Heeschen C, Sengenès C, Curat CA, Busse R, Bouloumie A. 2004. Improvement of postnatal neovascularization by human adipose tissue-derived stem cells. *Circulation* 110:349-355.
25. Mitchell JB, McIntosh K, Zvonic S, Garrett S, Floyd ZE, Kloster A, Di Halvorsen Y, Storms RW, Goh B, Kilroy G, Wu X, Gimble JM. 2006. Immunophenotype of human adipose-derived cells: temporal changes in stromal-associated and stem cell-associated markers. *Stem Cells* 24:376-385.
26. Mizuno H. 2009. Adipose-derived stem cells for tissue repair and regeneration: ten years of research and a literature review. *J Nippon Med Sch* 76: 56-66.
27. Morgan SM, Ainsworth BJ, Kanczler JM, Babister JC, Chaudhuri JB, Oreffo RO. 2009. Formation of a human-derived fat tissue layer in P(DL)LGA hollow fibre scaffolds for adipocyte tissue engineering. *Biomaterials* 30:1910-1917.
28. Natesan S, Baer DG, Walters TJ, Babu M, Christy RJ. 2010. Adipose-derived stem cell delivery into collagen gels using chitosan microspheres. *Tissue Eng Part A* 16:1369-84.
29. O'Brien FJ, Harley BA, Yannas IV, Gibson LJ. 2005. The effect of pore size on cell adhesion in collagen-GAG scaffolds. *Biomaterials* 26:433-41.
30. Romanov YA, Svintsitskaya VA, Smirnov VN. 2003. Searching for alternative sources of postnatal human mesenchymal stem cells: Candidate Msc-like cells from umbelical cord, *Stem Cells* 21:105-110.
31. Rosson GD, Magarakis M, Shridharani SM, Stapleton SM, Jacons LK, Manahan MA, Flores JJ. 2010. A review of the surgical management of breast cancer: Plastic reconstructive techniques and timing implications. *Ann Surg Oncol* 17:1890-900.
32. Roubelakis MG, Bitsika V, Zagoura D, Trohatou O, Pappa KI, Makridakis M, Antsaklis A, Vlahou A, Anagnostou NP. 2011. In vitro and in vivo properties of distinct populations of amniotic fluid mesenchymal progenitor cells. *J Cell Mol Med* 15:1896-913.
33. Rubin JP, Bennett JM, Doctor JS, Tebbets BM, Marra KG. 2007. Collagenous microbeads as a scaffold for tissue engineering with adipose-derived stem cells. *Plast Reconstr Surg* 120:414-24.
34. Schilling T, Nöth U, Klein-Hitpass L, Jakob F, Schütze N. 2007. Plasticity in adipogenesis and osteogenesis of human mesenchymal stem cells. *Molecular and Cellular Endocrinology* 271:1-17.
35. Spees JL, Olson SD, Whitney MJ, Prockop DJ. 2006. Mitochondrial transfer between cells can rescue aerobic respiration. *Proc Natl Acad Sci U S A* 103:1283–1288.

36. Stosich MS and Mao JJ. 2005. Adipose tissue engineering from human adult stem cells: clinical implications in plastic and reconstructive surgery. *Plast Reconstr Surg* 119:71-83.
37. Von Heimburg D, Kuberka M, Rendchen R, Hemmrich K, Rau G, Pallua N. 2003. Preadipocyte-loaded collagen scaffolds with enlarged pore size for improved soft tissue engineering. *Int J Artif Organs* 26:1064-76.
38. Von Heimburg D, Zachariah S, Heschel I, Kühling H, Schoof H, Hafemann B, Pallua N. 2001. Human preadipocytes seeded on freeze-dried collagen scaffolds investigated in vitro and in vivo. *Biomaterials* 22:429-38.
39. Wang S, Qu X, Zhao RC. 2011. Mesenchymal stem cells hold promise for regenerative medicine. *Front Med* 5:372-378.
40. Witkowska-Zimmy M and Walenko K. 2011. Stem cells from adipose tissue. *Cell Mol Biol Lett* 16:236-257.
41. Woodbury D, Schwarz EJ, Prockop DJ, Black IB. 2000. Adult rat and human bone marrow stromal cells differentiate into neurons. *J Neurosci Res* 61:364-70.
42. Yoshimura K, Shigeura T, Matsumoto D, Sato T, Takaki Y, Aiba-Kojima E, Sato K, Inoue K, Nagase T, Koshima I, Gonda K. 2006. Characterization of freshly isolated and cultured cells derived from the fatty and fluid portions of liposuction aspirates. *J Cell Physiol* 208:64-76.
43. Zech NM, Preisegger KH, Hollands P. 2011. Stem cell therapeutics - reality versus hype and hope. *J Assist Reprod Genet* 28:287-290.
44. Zuk PA, Zhu M, Ashjian P, De Ugarte JI, Mizuno H, Alfonso ZC, Fraser JK, Benhaim P, Hedrick MH. 2002. Human adipose tissue is a source of multipotent stem cells. *Mol Biol Cell* 13:4279-4295.
45. Zuk PA, Zhu M, Mizuno H, Huang J, Futrell JW, Katz AJ, Benhaim P, Lorenz HP, Hedrick MH. 2001. Multilineage cells from human adipose tissue: implications for cell-based therapies. *Tissue Eng* 7:211-28.

## FIGURE LEGENDS

**Fig 1.** Representative image of flow cytometric analyses performed at day 0, 15, and 30 of culture.

(A,B) At day 0, a significant percentage of ASCs are clearly positive for stromal markers, including CD117 (5%), CD34 (7%); for mesenchymal markers, including CD29 (72%), CD105 (5%), CD34 (25%), CD90 (30%) and for endothelial markers, including CD31 (6%), CD133 (8%), CD44/CD54 (5%), and Flk-1 (45%); (C) Sorting image for CD34<sup>+</sup>CD90<sup>+</sup>; (D) At day 15 co-expression of CD34 and CD90 markers was 80%; (E) After 30 days of culture, co-expression of CD34 and CD90 was 8%, while only CD90 expression was 80%.

**Fig 2.** Representative image showing growth curves of CD34<sup>+</sup>/CD90<sup>+</sup> cells with respect to CD34<sup>-</sup>/CD90<sup>-</sup> and unsorted cells displaying a different mean doubling time.

**Fig 3.** Image showing (A) unsorted cells in adipogenic medium, exhibiting the morphology of fibroblasts (arrowheads), whereas other cells exhibiting the morphology of multi-vacuolar adipocytes (arrows) (Scale bar: 150 μm); (B) some unsorted cells in adipogenic medium, displaying positivity for adiponectin by immunohistochemistry (Scale bar: 150 μm); (C) CD34<sup>+</sup>/CD90<sup>+</sup> ASCs in adipogenic medium, exhibiting the characteristic adipocyte morphology (Scale bar: 250μm); (D) CD34<sup>+</sup>/CD90<sup>+</sup> ASCs in adipogenic medium, displaying positivity for adiponectin by immunohistochemistry (Scale bar: 250μm); (E) CD34<sup>+</sup>/CD90<sup>+</sup> ASCs in adipogenic medium, showing positivity for PPAR-γ by immunohistochemistry (Scale bar: 150μm). (F) differentiated endothelial cells that, after day 7 of culture in presence of methylcellulose, formed a rather extended intercellular tube network (Scale bar: 250μm); (G) flow cytometry demonstrating that a significant number of ASCs are positive for endothelial markers, including CD90 (97%), CD44 (90%), CD54 (90%), VEGF (70%), CD133 (18%), and Flk-1 (60%). Control FITC-conjugated and PE-conjugated isotypes were in green.

**Fig. 4.** Figure showing the viability of sorted ASCs in (A) controls (Scale bar: 100μm) and (B) when loaded onto the scaffold (Scale bar: 100μm). In both cases Calcein staining demonstrates a high viability, also after adherence to the scaffold in (B). In order to evaluate cell proliferation,

constructs were analyzed by inverted microscope after 7 (C) and 14 day (D) and by histology after 7 (E) and 14 day (F) (Scale bar: 250 $\mu$ m). Light-inverted microscopy and H&E staining on serial sections of ASCs seeded onto the Gingistat collagen sponge demonstrated that the cells were homogeneously distributed throughout the whole sponge. Moreover, at day 14, either a strong spreading or ASCs proliferation was observed with respect to day 7 (arrowheads indicate the scaffold and the arrows indicate the cells).

**Fig. 5.** Figure showing in (A) sorted ASCs/collagen construct before implantation; (B) sorted ASCs/collagen construct injected into nude mice and withdrawn after 30 days of implantation; (C) sorted ASCs/Collagen construct explanted from a nude mouse; (D) Collagen scaffold, grafted without cells (used as control) which was not detectable on the dorsum of mice, because completely reabsorbed.

**Fig. 6.** Figure showing: (A) H&E stain displaying that, within the unsorted ASCs/collagen constructs, stem cells only differentiated towards adipocytes (arrowheads indicate the scaffold and the arrows indicate the adipocytes) (Scale bar: 150 $\mu$ m); (B) H&E staining displaying that, within the sorted ASCs/collagen constructs, stem cells differentiated towards adipocytes (Scale bar 250 $\mu$ m); (C) H&E staining shows that fat cell are often surrounded by a network of loose connective tissue (Scale bar 250 $\mu$ m); (D) Collagenous fibres predominated and constituted the bulk of the loose connective tissue (Mallory's Trichrome staining) (Scale bar: 250 $\mu$ m); (E) Class I HLA positive staining clearly demonstrated the human origin of the adipose tissue obtained excluding any murine contamination (Scale bar 100 $\mu$ m); (F) Immunofluorescence staining for adiponectin (Scale bar 100 $\mu$ m); (G) Immunofluorescence staining for Glut-4 (Scale bar: 100 $\mu$ m); (H) Immunofluorescence staining for PPAR- $\gamma$  (Scale bar: 250 $\mu$ m); (I) Immunofluorescence staining for Collagen Type I (Scale bar 250 $\mu$ m); (J) Immunofluorescence staining for VEGF (arrowheads indicate the capillary-like cells network). (Scale bar: 250 $\mu$ m);

**Fig. 7.** RT-PCR analyses. Figure showing adipose differentiation-associated genes in sorted ASCs grown on collagen sponge, after 30 days from implantation. Analyses revealed that PPAR- $\gamma$  and

adiponectin expressions as well as VEGF were highly expressed, confirming the adipose tissue formation with neo-angiogenesis.

Table 1. *In vivo* construct implantation protocol

	Right side	Left side	Scaffold size	Cell number
Nude mouse 1	unsorted ASCs/Collagen	ASCs CD34 <sup>+</sup> CD90 <sup>+</sup> /Collagen	10x10 mm	2x10 <sup>5</sup> cells
Nude mouse 2	unsorted ASCs/Collagen	ASCs CD34 <sup>+</sup> CD90 <sup>+</sup> /Collagen	10x10 mm	2x10 <sup>5</sup> cells
Nude mouse 3	unsorted ASCs/Collagen	ASCs CD34 <sup>+</sup> CD90 <sup>+</sup> /Collagen	10x10 mm	2x10 <sup>5</sup> cells
Nude mouse 4	unsorted ASCs/Collagen	ASCs CD34 <sup>+</sup> CD90 <sup>+</sup> /Collagen	10x10 mm	2x10 <sup>5</sup> cells
Nude mouse 5	unsorted ASCs/Collagen	ASCs CD34 <sup>+</sup> CD90 <sup>+</sup> /Collagen	10x10 mm	2x10 <sup>5</sup> cells
Nude mouse 6	unsorted ASCs	ASCs CD34 <sup>+</sup> CD90 <sup>+</sup>	0	2x10 <sup>5</sup> cells
Nude mouse 7	unsorted ASCs	ASCs CD34 <sup>+</sup> CD90 <sup>+</sup>	0	2x10 <sup>5</sup> cells
Nude mouse 8	Collagen	Collagen	10x10 mm	0
Nude mouse 9	Collagen	Collagen	10x10 mm	0

Table 2. The *in vivo* construct explantation

	Right side	Tissue volume (mm <sup>3</sup> )	New tissue formed	Left side	Tissue volume (mm <sup>3</sup> )	New tissue formed
Nude mouse 1	unsorted ASCs/Collagen	85.2 ± 1,1	Adipose tissue	ASCs CD34 <sup>+</sup> CD90 <sup>+</sup> /Collagen	152.3 ± 1,4	Adipose and loose connective
Nude mouse 2	unsorted ASCs/Collagen	129 ± 0,8	Adipose tissue	ASCs CD34 <sup>+</sup> CD90 <sup>+</sup> /Collagen	102.5 ± 0,6	Adipose and loose connective
Nude mouse 3	unsorted ASCs/Collagen	135 ± 1,5	Adipose tissue	ASCs CD34 <sup>+</sup> CD90 <sup>+</sup> /Collagen	95.6 ± 1,1	Adipose and loose connective
Nude mouse 4	unsorted ASCs/Collagen	100.8 ± 0,3	Adipose tissue	ASCs CD34 <sup>+</sup> CD90 <sup>+</sup> /Collagen	132.8 ± 0,8	Adipose and loose connective
Nude mouse 5	unsorted ASCs/Collagen	147.6 ± 0,5	Adipose tissue	ASCs CD34 <sup>+</sup> CD90 <sup>+</sup> /Collagen	146.5 ± 1,2	Adipose and loose connective
Nude mouse 6	unsorted ASCs	reabsorbed	reabsorbed	ASCs CD34 <sup>+</sup> CD90 <sup>+</sup>	reabsorbed	reabsorbed
Nude mouse 7	unsorted ASCs	reabsorbed	reabsorbed	ASCs CD34 <sup>+</sup> CD90 <sup>+</sup>	reabsorbed	reabsorbed
Nude mouse 8	Collagen	reabsorbed	reabsorbed	Collagen	reabsorbed	reabsorbed
Nude mouse 9	Collagen	reabsorbed	reabsorbed	Collagen	reabsorbed	reabsorbed

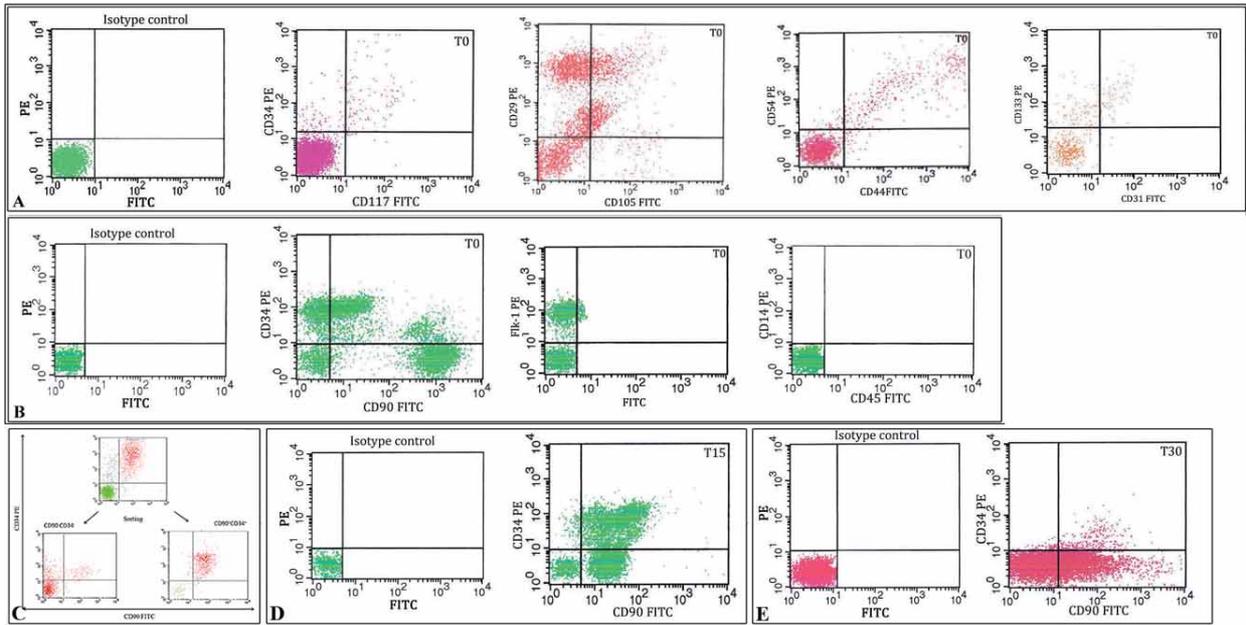


Figure 1

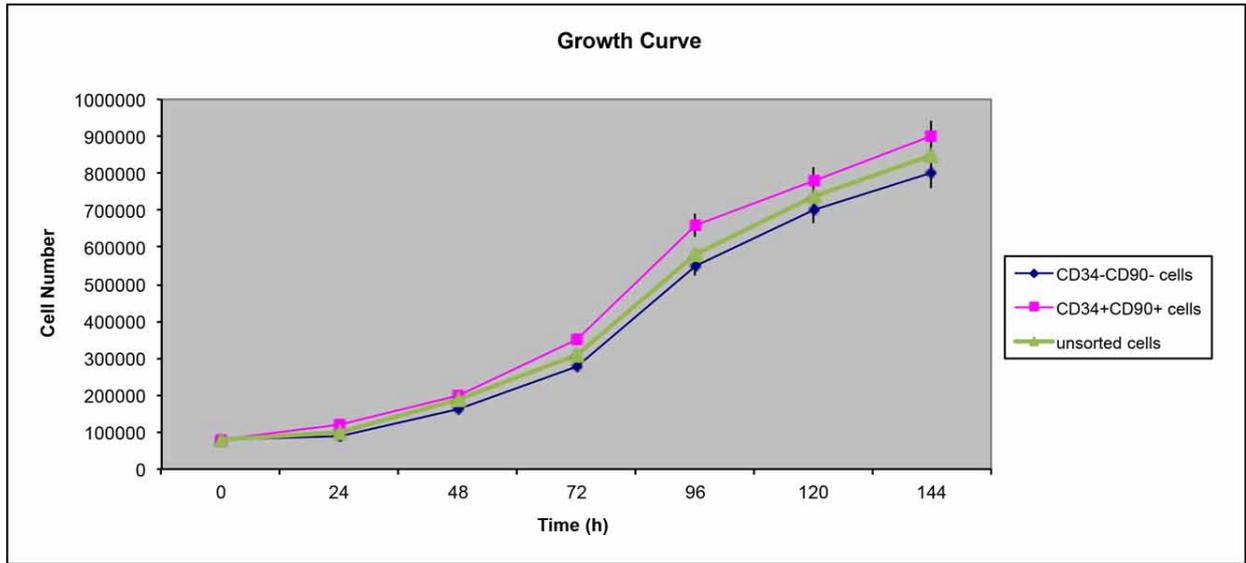


Figure 2

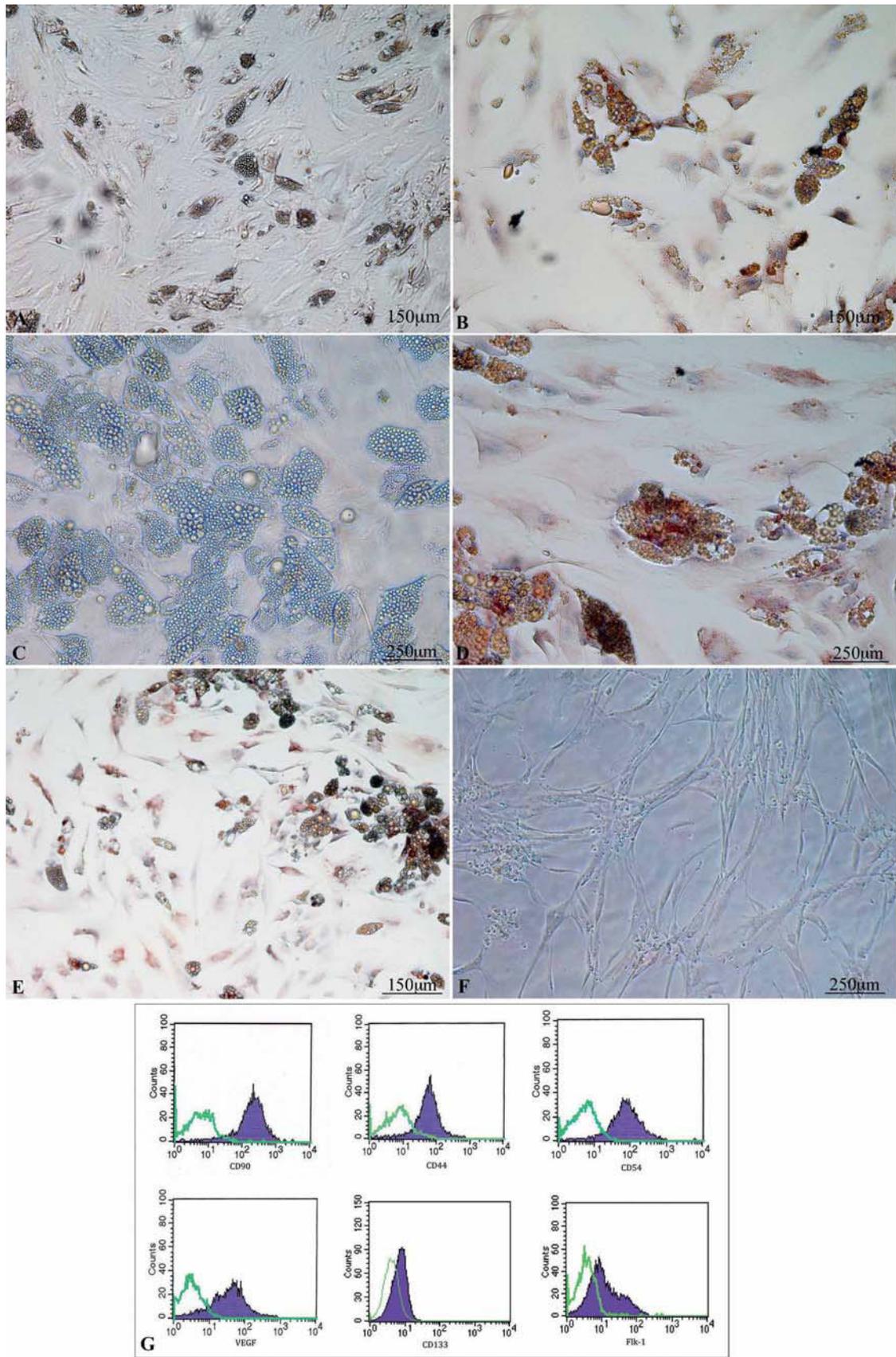


Figure 3

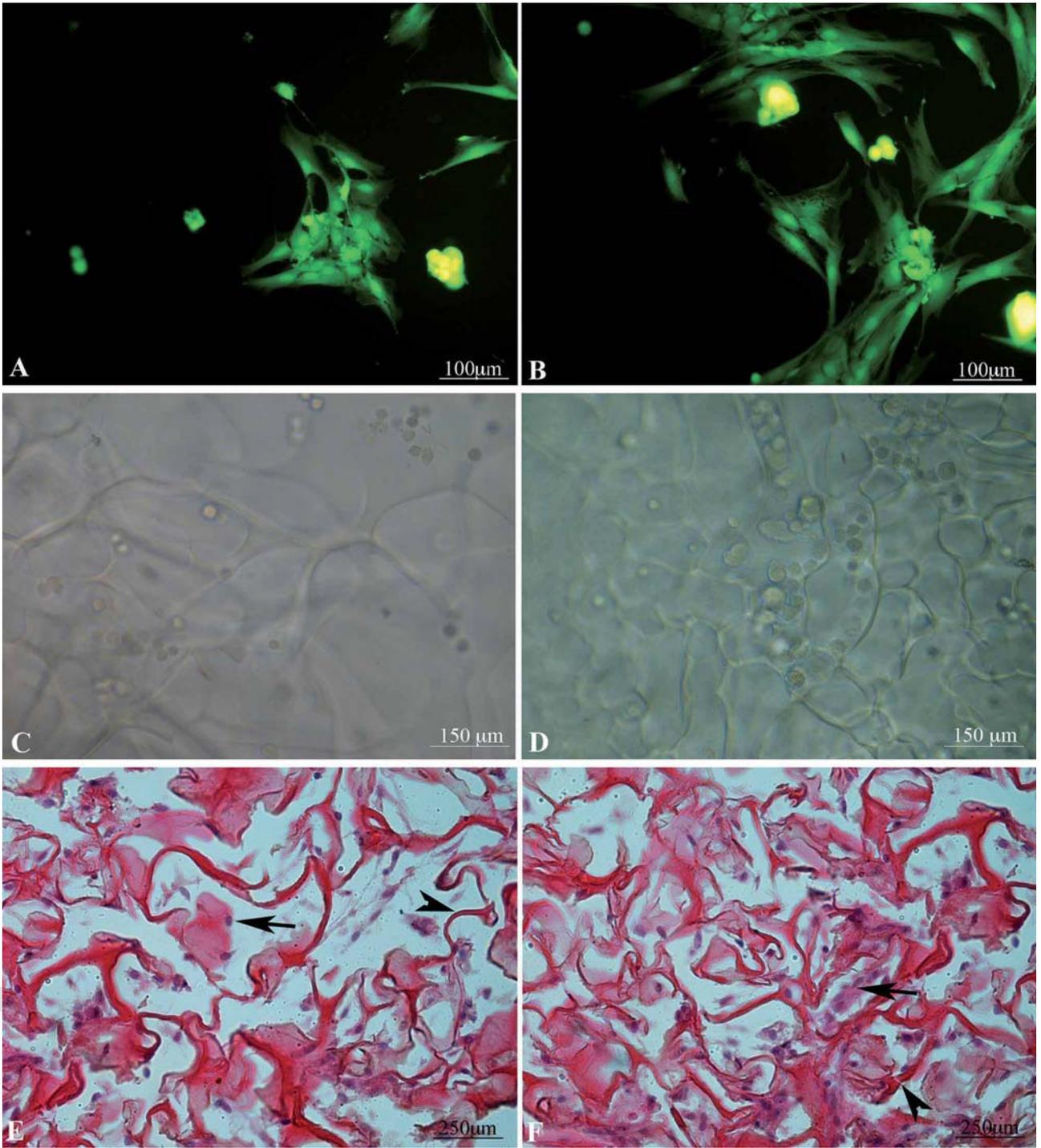


Figure 4

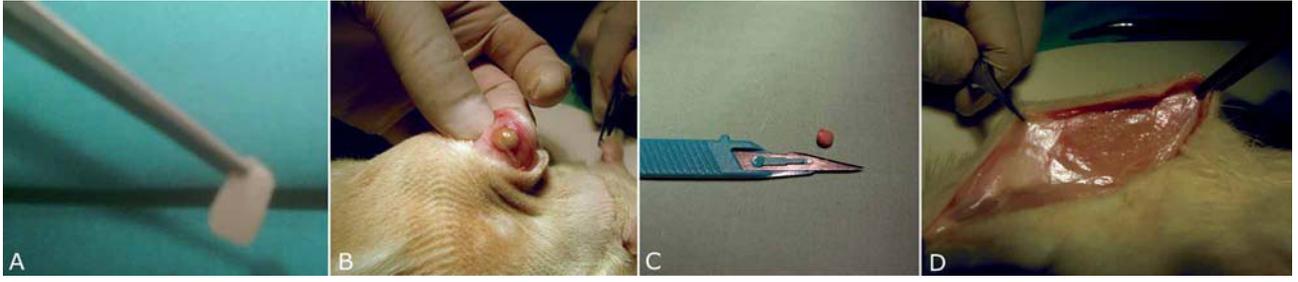


Figure 5

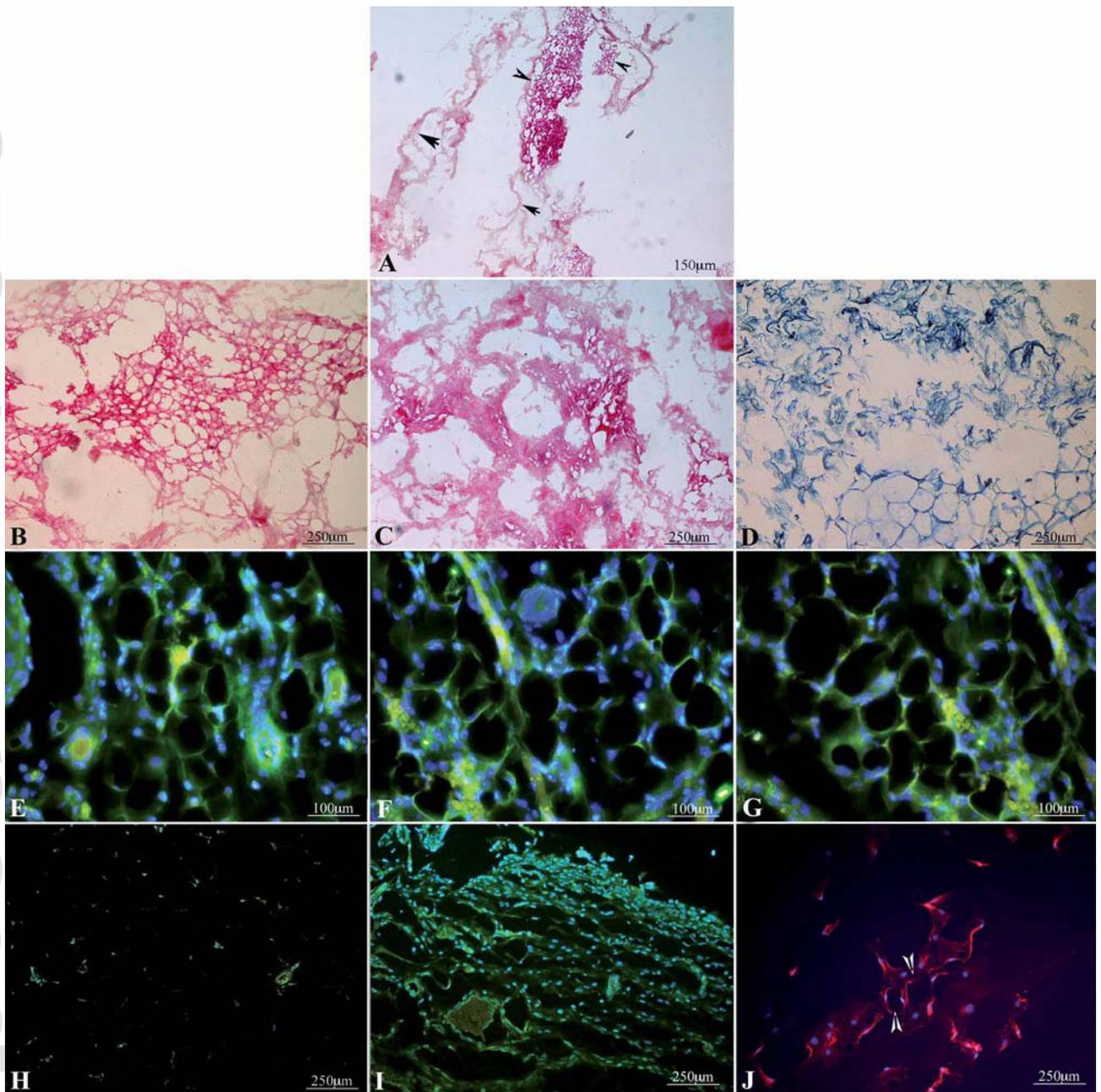


Figure 6

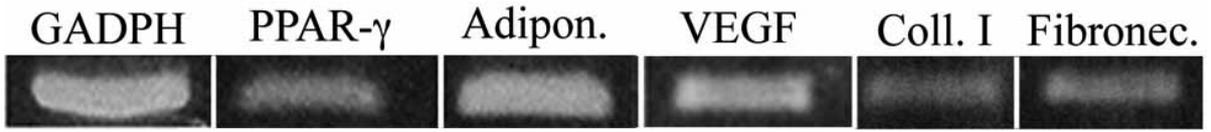


Figure 7