Human Blood and Cardiac Stem Cells Synergize to Enhance Cardiac Repair When Cotransplanted Into Ischemic Myocardium

Nicholas Latham, BSc; Bin Ye, MSc; Robyn Jackson, BSc; Bu-Khanh Lam, MD; Drew Kuraitis, BSc; Marc Ruel, MD; Erik J. Suuronen, PhD; Duncan J. Stewart, MD; Darryl R. Davis, MD

Background—Blood-derived circulatory angiogenic cells (CACs) and resident cardiac stem cells (CSCs) have both been shown to improve cardiac function after myocardial infarction. The superiority of either cell type has long been an area of speculation with no definitive head-to-head trial. In this study, we compared the effect of human CACs and CSCs, alone or in combination, on myocardial function in an immunodeficient mouse model of myocardial infarction.

Methods and Results—CACs and CSCs were cultured from left atrial appendages and blood samples obtained from patients undergoing clinically indicated heart surgery. CACs expressed a broader cytokine profile than CSCs, with 3 cytokines in common. Coculture of CACs and CSCs further enhanced the production of stromal cell–derived factor-1α and vascular endothelial growth factor (P≤0.05). Conditioned media promoted equivalent vascular networks and CAC recruitment with superior effects using cocultured conditioned media. Intramyocardial injection of CACs or CSCs alone improved myocardial function and reduced scar burdens when injected 1 week after myocardial infarction (P≤0.05 versus negative controls). Cotransplantation of CACs and CSCs together improved myocardial function and reduced scar burdens to a greater extent than either stem cell therapy alone (P≤0.05 versus CAC or CSC injection alone).

Conclusions—CACs and CSCs provide unique paracrine repertoires with equivalent effects on angiogenesis, stem cell migration, and myocardial repair. Combination therapy with both cell types synergistically improves postinfarct myocardial function greater than either therapy alone. This synergy is likely mediated by the complimentary paracrine signatures that promote revascularization and the growth of new myocardium. (Circulation. 2013;128[suppl 1]:S105-S112.)

Key Words: blood cells ■ heart failure ■ myocardial infarction ■ stem cells

The search for cell products capable of myocardial repair has yielded a variety of candidates. In the past 10 years, circulatory angiogenic cells (CACs or early outgrowth endothelial progenitor cells) have emerged as the most promising subtype of blood-derived stem cells for myocardial repair, given their capacity to form new blood vessels (vasculogenesis) while stimulating existing blood vessels to expand (angiogenesis) through paracrine stimulation. As a result, this promising cell candidate is under investigation in a variety of clinical trials.2,3

Cardiac stem cells (CSCs) have also emerged as an attractive cell candidate for myocardial repair because they offer an autologous cell product genetically preprogrammed to form heart tissue.4 When injected into animal models of cardiac damage, CSCs differentiate into new working heart tissue and provide functional improvements.5 Data from phase 1 studies confirm preclinical experience and have provided the impetus for recently started phase 2 trials.6,7 But despite clear evidence of benefit after cell transplantation, long-term retention of cells is modest (<5% at 3 weeks), regardless of persistent functional benefits.8 These data hint that a sizable portion of myocardial repair is mediated by paracrine stimulation of endogenous repair or myocardial salvage mechanisms.9

Although both CAC and CSC transplantation provide myocardial repair, speculation remains as to which cell source is superior. To provide a real-world context, this study will compare the effect of primary cultured human CACs and CSCs on myocardial function after delayed injection into an immunodeficient mouse model of myocardial ischemia. Both cell sources will be cultured from cardiac patients undergoing clinically indicated procedures and reflect the target population currently enrolled in clinical trials. The fundamental mechanisms underlying cell-mediated benefits will be contrasted by comparing angiogenic and paracrine profiles. Given
the disparate ontogeny and possible mechanism of benefit, the effects of combination therapy will be explored to discover whether coadministration of both cell types is detrimental, ineffectual, or synergistic.

**Methods**

See the expanded methods section (online-only Data Supplement) for more details.

**Cell Culture and Conditioned Media**

Human CACs and CSCs were cultured from tissue samples obtained from patients undergoing clinically indicated heart surgery after informed consent. All protocols were approved by the University of Ottawa Heart Institute Research Ethics Board. CSCs were cultured using standard culture techniques described previously. Normal human dermal fibroblasts (NHDFs) and human umbilical vein endothelial cells were cultured according to the manufacturer’s directions. Conditioned media was obtained from CSCs, CACs, and NHDFs after 48 hours of culture in low serum basal media under hypoxic conditions (1% oxygen) to simulate the environment of the infarcted myocardium. Cocultures of CACs and CSCs were seeded at 3 confluency ratios (CSClow/CAChigh; CSChigh/CAClow; CSChigh/CAChigh) to examine the relationship between CACs and CSCs under coculture conditions.

**In Vitro Cytokine Expression**

Cytokine secretion in conditioned media was screened using a custom protein array (RayBiotech, USA) according to the manufacturer’s directions. Cytokines of interest were confirmed using ELISA. All immunosorbent measures were normalized to the protein content and media volume.

**In Vitro Angiogenic Differentiation and Cell Migration**

The capacity of CACs and CSCs to stimulate angiogenic growth was assessed using a growth factor-depleted matrigel assay (Millipore) in accordance with the manufacturer’s instructions. Six random fields were analyzed, and cumulative tubular growth was determined. The ability of conditioned media to recruit CACs was assessed using transwell plates (3.0 μm pores; Corning). CACs that had successfully migrated through the polycarbonate membrane were fixed and stained with 4',6-diamidino-2-phenylindole (Sigma-Aldrich). Fluorescent microscopy was used to determine the average number of cells per random field.

**Myocardial Infarction, Cell Injection, and Functional Evaluation**

Myocardial infarctions were performed in male NOD-SCID mice by permanent ligation of the left anterior descending (LAD) coronary artery. Seven days after ligation, stem cells and controls were injected into the myocardium along the infarct border and at the cardiac apex using transthoracic echocardiographic guidance (VisualSonics). Left ventricular ejection fraction was evaluated 21 and 28 days after LAD ligation to assess the functional effects of each cell therapy. Long-term effects of cell therapy were evaluated in a subset of mice from each group (n=4). All functional evaluations were conducted and analyzed by investigators blinded to the animal’s treatment group. After the last assessment of myocardial function, the mice were euthanized and hearts excised for histology or quantitative polymerase chain reaction (PCR) analysis.

**Quantitative PCR Analysis**

Myocardial retention of transplanted cells was assessed in a subset of mice (n=3/group) using quantitative PCR for noncoding human alu repeats. Left ventricular genomic DNA was extracted, and quantitative PCR was performed with transcript-specific hydrolysis primer probes.

**Histology**

Tissue viability was assessed after staining with Masson trichrome (Invitrogen). Stem cell engraftment, capillary density, and differentiation were assessed through immunohistochemistry. For these measures, 3 sections were analyzed per animal and averaged with ≥3 animals per group.

**Statistical Analysis**

All data are presented as mean±SEM. To determine whether differences existed within groups, data were analyzed by a 1-way or repeated-measures ANOVA (SPSS version 20.0.0); if such differences existed, Bonferroni-corrected t test was used to determine the groups with the differences. In all cases, variances were assumed to be equal, and normality was confirmed before further post hoc testing.
Differences in categorical measures were analyzed using a \( \chi^2 \) test. A final value of \( P \leq 0.05 \) was considered significant for all analyses.

**Results**

**Baseline Demographics**

Fifty-three patients (69% men; age, 68±2 years; body mass index, 29±1 kg/m\(^2\); Table I in the online-only Data Supplement) were enrolled in the study. All patients had a history of stable cardiac disease with numerous cardiovascular risk factors, including diabetes mellitus (37%), hypertension (74%), and dyslipidemia (65%). The majority of patients had a history of coronary artery disease (75%), myocardial infarction (22%), valvular heart disease (31%), and congestive heart failure (31%). The majority of patients underwent elective cardiac surgery for coronary bypass alone (65%), with the remainder undergoing valve repair/replacement alone (25%) or coronary bypass with valve repair/replacement (10%). No patient had experienced an acute coronary syndrome or admission for congestive heart failure for 6 months before sample collection. All patients were on stable cardiac medications, including angiotensin-converting enzyme inhibitors and angiotensin receptor blockers (82%), antiplatelet therapy (75%), \( \beta \)-blockers (69%), and statins (61%) for 26 months before surgery. Although the baseline clinical characteristics of the patients were similar, notable exceptions included a tendency for better renal function (1.2±0.1 versus 0.9±0.1 mL/min; \( P \leq 0.05 \)) and worse chronic stable angina (Canadian Cardiovascular Society class 1.2±0.1 versus 0.3±0.2; \( P \leq 0.05 \)) in patients who donated samples for the in vivo study.

Atrial appendage specimens were collected at the time of cardiac surgery and began processing within 1 hour of harvest. To provide an unbiased comparison of CAC and CSC efficacy, blood samples for in vitro testing were collected at the time of cardiac surgery (Figure 1). In deference to a clinically translatable protocol and the different times required for stem cell culture (6 versus 14 days), blood samples for in vivo testing were collected 8 days after cardiac surgery. Flow cytometry of representative collections of both cell types demonstrated characteristic proportions of CAC and CSC identity markers (Figure I in the online-only Data Supplement). Age and other comorbidities were not found to influence overall culture yield.

**Figure 2.** Growth factors produced by circulatory angiogenic cells (CACs), cardiac stem cells (CSCs), and normal human dermal fibroblasts (NHDFs) under hypoxic culture conditions. A, Representative images of the custom protein array used to screen conditioned media from CACs, CSCs, and NHDFs. Densitometry values were run in duplicates on the same array (Figure II in the online-only Data Supplement). B, Densitometry analysis of growth factors produced by CACs (n=6) and CSCs (n=8) compared with NHDFs (n=7). *P \leq 0.05 for CSCs vs NHDF; †P \leq 0.05 for CACs vs NHDF. AR indicates androgen receptor; bFGF, basic fibroblast growth factor; BMP, bone morphogenetic protein; EGF, epidermal growth factor; FGF, fibroblast growth factor; GCSF, granulocyte colony-stimulating factor; GM-CSF, granulocyte macrophage colony-stimulating factor; HB-EFG, heparin binding epidermal growth factor; ICAM, intracellular adhesion molecule; IFN, interferon; IGFBP, insulin-like growth factor-binding protein; IGF, insulin-like growth factor; IL, interleukin; MIP, macrophage inflammatory protein; PDGF, platelet-derived growth factor; PECAM, platelet endothelial cell adhesion molecule; SCF, stem cell factor; SDF, stromal cell-derived factor; TGF, transforming growth factor; TNF, tumor necrosis factor; VCAM, vascular cell adhesion molecule; HGF, hepatocyte growth factor; TIMP, tissue inhibitor of metalloproteinase; and VEGF, vascular endothelial growth factor.
Human CACs Express a Broader Cytokine Profile Than Human CSCs

The paracrine profile of human CSCs, CACs, and NHDFs was screened using conditioned media with a custom protein array. This array returned a proportional fluorescent signal for the 59 cytokines tested with 2 technical repeats (Figure II in the online-only Data Supplement). Figure 2 demonstrates 3 representative blots from human CSCs, CACs and NHDFs. As shown in Figure 2B, both CACs and CSCs produced a large number of growth factors in excess to NHDF (36 and 5 cytokines; \( P \leq 0.05 \) versus cytokine levels detected within NHDF-conditioned media). Interestingly, the paracrine profile of CACs was significantly broader than CSCs (\( \chi^2 \) value, 3.93; \( P \leq 0.05 \) versus the expected frequency of cytokines elevated in stem cell--conditioned media), with rare instances of the same growth factor being overexpressed by both cell types (angiopoietin-1, hepatocyte growth factor, and vascular endothelial growth factor).

Confirmatory ELISA analysis was performed on select cytokines based on high levels of expression or literature supporting a key role in postinfarct repair (Figure 3). These assays confirmed that CSCs produced greater amounts of angiogenin, hepatocyte growth factor, interleukin-6, stromal cell--derived factor-1\( \alpha \), and vascular endothelial growth factor, whereas CACs produced greater amounts of epidermal growth factor. The possibility that different combinations of cell types may interact to influence growth factor secretion was analyzed using cocultures at different confluency ratios. These combination cocultures corresponded to half the number of cells used in either single stem cell system (CSC\( \text{norm} \)/CAC\( \text{high} \) 5.0\( \times \)10\(^6\)/1.5\( \times \)10\(^7\); CSC\( \text{norm} \)/CAC\( \text{high} \) 1.0\( \times \)10\(^7\)/7.5\( \times \)10\(^5\); CSC\( \text{norm} \)/CAC\( \text{high} \) 1.0\( \times \)10\(^7\)/1.5\( \times \)10\(^6\)). Combination culture did not provide additional production of epidermal growth factor and hepatocyte growth factor under all 3 coculture conditions (\( P \leq 0.05 \) versus single cultures), whereas angiogenin, stromal cell--derived factor-1\( \alpha \), and vascular endothelial growth factor were all produced in an incremental fashion (\( P \leq 0.05 \) versus single cultures). These data suggest that important costimulation occurs between the different cell types, which may increase the potency of combination therapy when CACs and CSCs are administered together.

Human CACs and CSCs Increase Angiogenesis and Cell Migration

The capacity of human CACs and CSCs to form blood vessels was assessed by exposing umbilical vein endothelial cells to stem cell--conditioned media within a growth factor--depleted matrigel assay (Figure 4). Media conditioned from CAC and CSC cultures stimulated vessel formation to a similar extent (\( P = \text{ns} \)). Conditioned media from cocultures demonstrated an additive effect with more tubule formation (\( P \leq 0.05 \)). Conditioned media from CAC and CSC cultures attracted CACs to a similar extent (\( P = \text{ns} \)), whereas conditioned media from CSC/CAC cocultures showed a greater capacity to...
attract CACs than either single culture alone ($P \leq 0.05$). These results suggest that CSC and CACs have a similar capacity to support angiogenesis, whereas coculture further enhances this effect.

**Human CACs and CSCs Provide Equivalent Myocardial Repair With Superior Benefits Using Combination Therapy**

The effect of human CACs and CSCs alone or in combination was assessed after intramyocardial injection into an immuno-deficient mouse model of myocardial ischemia. As shown in Figure 5A, animals treated with CACs or CSCs alone had a greater ejection fraction (37±2% and 36±2%, respectively) 3 weeks after LAD ligation than animals treated with NHDF or PBS (22±2% and 23±1%, respectively; $P \leq 0.05$). These benefits were maintained in both individual treatment groups 3 months after LAD ligation (37±2% and 36±2%, respectively; Table II in the online-only Data Supplement). In a manner consistent with the in vitro data, cotransplantation of CACs and CSCs provided greater myocardial repair 3 weeks after LAD ligation compared with injection with either cell type alone (Figure 5A). Long-term data from a subset of mice (n=4) suggest that these effects are sustained ($P$=ns, +28 days versus +16 weeks after LAD ligation left ventricular ejection fraction) compared with the progressive marked decline in the PBS treatment group (+16-week ejection fraction $P \leq 0.05$ compared with CAC or CSC-alone treatment; Figure IIIA in the online-only Data Supplement).

These functional benefits occurred despite modest retention of injected cells (Figure 5B). Furthermore, despite equivalent degrees of myocardial repair, fewer CSCs were found 21 days after intramyocardial injection compared with

---

**Figure 4.** Proangiogenic effects of circulatory angiogenic cells (CACs) and cardiac stem cells (CSCs). **A**, Representative images of matrigel-cultured umbilical vein endothelial cells (HUVECs) exposed to conditioned media from CACs, CSCs, normal human dermal fibroblast (NHDF), and serum-free controls. **B**, Cumulative tubular length analysis demonstrates greater vessel formation in stem cell–conditioned media compared with the negative cellular control. Conditioned media from the coculture of CACs and CSCs further enhanced HUVEC vessel formation. **C**, Representative images of CACs after migration through the transwell filter when exposed to conditioned media from CACs, CSCs, NHDF, and CSC/CAC cocultures. **D**, Analysis of the number of cells that migrated through the transwell filter after exposure to conditioned media and normalized to unbiased vascular endothelial growth factor (VEGF)–alone stimulation. *$P$≤0.05; **$P$≤0.05 compared with all other cell cultures.
CACs (0.5±0.1% versus 3.6±1.1%; P<0.05). Combination therapy with both CACs and CSCs did not enhance engraftment or cell retention 21 days after injection (P=ns), although superior effects on myocardial repair were observed. Long-term engraftment data demonstrated that although human CSCs continued to persist in the mouse myocardium CAC retention dwindled to comparable numbers by 16 weeks after transplantation (Figure IIIB in the online-only Data Supplement). Taken together, these data hint that the benefits observed with first-generation CAC and CSC products are independent of long-term myocardial retention and reflect the contribution of growth factors delivered in the first weeks after cell injection.

Transplantation of Human CAC and CSCs Reduce Ventricular Scar Burden With Superior Effects Using Combination Therapy

Scar formation and tissue viability within the infarct zone were analyzed using Masson trichrome–stained sections 21 days after stem cell injection (Figure 6). Both CAC and CSC transplantation reduced scar formation compared with PBS-treated animals (16.7±1.0% and 13.9±0.8% versus 23.0±1.6%, respectively; P≤0.05). CSC therapy alone prevented scar formation to a greater degree compared with CAC-treated animals (P≤0.05), despite equivalent effects on myocardial function. Transplantation of both cell types in combination reduced ventricular scar burden to a greater degree compared with either cell type alone (8.9±1.0%; P≤0.05). Cell-mediated effects on ventricular scarring were sustained over long-term follow-up (Figure IV in the online-only Data Supplement). CAC- and CSC-treated animals demonstrated a significantly higher capillary density within the peri-infarct region compared with PBS-treated controls (27.8±3.2% and 22.2.9±2.1% versus 15.1±2.2%, respectively; P≤0.05). Although single-cell therapies yielded comparable capillary densities, combination cell therapy improved capillary density over all treatment groups (42.6±2.2%; P≤0.05; Figure V in the online-only Data Supplement).

Small Clusters of Differentiated Human Cells Persist Within the Infarct and Peri-infarct Regions

To evaluate stem cell engraftment and differentiation, histological sections were labeled with human nuclear antigen in conjunction with cardiac lineage markers (smooth muscle [α-smooth muscle actin], vascular [Von Willebrand factor], and myocyte [cardiac troponin T]; Figure 7). Small clusters of human cells were identified 21 days after stem cell transplantation in each treatment group within the peri-infarct region, as well as the infarct itself. This indicated that each stem cell treatment provided cells capable of engrafting and differentiating into functional cells within the damaged myocardium, albeit at a modest degree. Animals transplanted with human
CACs alone had only human cells of vascular identity found on follow-up histology (Figure VI in the online-only Data Supplement). In contrast, animals treated with either CSCs alone or combination of CACs+CSCs had human cells of all 3 lineages found, demonstrating the inherent multilineage potential of CSCs.

Discussion

This study demonstrates that the 2 leading clinical candidates for cell-mediated cardiac repair provide unique paracrine repertoires with equivalent effects on angiogenesis, stem cell migration, and myocardial repair. Combination therapy with both first-generation cell products synergistically improved postinfarct myocardial function greater than either therapy alone. This synergy is likely mediated by the complimentary paracrine signatures that promote revascularization, tissue salvage, and the growth of new myocardium.

Given that these 2 cell types represent the 2 leading autologous cell sources in phase 1 and 2 cardiac repair trials, we chose to contrast cell products from patients undergoing clinically indicated cardiac surgery, precisely the same patients enrolled in current trials and in need of cellular cardiomyoplasty in the future. To provide an unbiased comparison of stem cell potency, in vitro experiments contrasted CACs and CSCs acquired at the time of cardiac surgery. The in vivo study was designed such that atrial appendages were acquired at the time of cardiac surgery, whereas CAC blood draws were performed 9 days after surgery. This strategy reflects the logistical realities inherent in cell culture and the administration of an autologous cell product soon after myocardial infarction. Although this approach may have favored CAC function and viability, this bias is tempered by the observation that (1) the effects of vascular damage on CAC function are very transient, (2) the results observed in both the in vivo and in vitro experiments are consistent, and (3) the phenotypic profile of both injected cells sources is consistent with those used in clinical trials (Figure I in the online-only Data Supplement ). The CSC product used in this study represents the ultimate simplification of CSC culture before antigenic selection or subculture. This product has been shown to provide equivalent repair to established cell products with hints of superiority.

The current work extends previous studies by providing the first unambiguous head-to-head comparison of autologous CACs and CSCs from clinical patients. Although earlier work from our laboratory demonstrated that expanded populations of CSCs from human cardiac samples secrete significant quantities of vascular endothelial growth factor, insulin-like growth factor-1, and hepatocyte growth factor, we used a wide-ranging cytokine protein array to show that CSCs also secrete significant amounts of angiopoietin-1, angiogenin, and interleukin-6. Interestingly, the CSC paracrine signature had few overlap cytokines with the more elaborate CAC paracrine profile. Despite these differences, the angiogenic response to conditioned media was similar between the 2 cells types, suggesting this plays an important role in cardiac repair. Although CSCs differentiated more readily into a cardiac phenotype, effects on cardiac repair were equivalent. This result may be explained by the PCR engraftment data demonstrating modest long-term retention (<1% of transplanted cells detected 3 weeks after injection). Despite this finding, long-term (+16 weeks after LAD ligation) benefits are sustained notwithstanding low numbers of injected cells. These data suggest that the benefits after CAC and CSC transplantation occur immediately on injection, and the persistence of large numbers of injected cells may not be necessary for sustained effects on myocardial repair, but this requires further testing to fully validate.

Given the number of CSC candidates of dissimilar ontogeny, it follows that treatment with complimentary cell types may provide synergistic benefits. The limited published data to date support this notion because treatment with nonclinically relevant cell types (ie, angiogenic cells+skeletal myoblasts or epicardium-derived cells+cardiac progenitor cells) demonstrates superior effects when combination cell products are injected. This report confirms this notion and provides
direct evidence that application of the 2 leading preclinical agents for myocardial repair provides synergistic benefits when applied after myocardial infarction.

Sources of Funding
This work was supported by the Canadian Institutes of Health Research (Operating Grant 229694). Dr Davis is funded by the Canadian Institutes of Health Research (Clinician Scientist Award).

Disclosures
None.

References
Human Blood and Cardiac Stem Cells Synergize to Enhance Cardiac Repair When Cotransplanted Into Ischemic Myocardium
Nicholas Latham, Bin Ye, Robyn Jackson, Bu-Khanh Lam, Drew Kuraitis, Marc Ruel, Erik J. Suuronen, Duncan J. Stewart and Darryl R. Davis

_Circulation_. 2013;128:S105-S112
doi: 10.1161/CIRCULATIONAHA.112.000374

_Circulation_ is published by the American Heart Association, 7272 Greenville Avenue, Dallas, TX 75231
Copyright © 2013 American Heart Association, Inc. All rights reserved.
Print ISSN: 0009-7322. Online ISSN: 1524-4539

The online version of this article, along with updated information and services, is located on the World Wide Web at:
http://circ.ahajournals.org/content/128/11_suppl_1/S105

Data Supplement (unedited) at:
http://circ.ahajournals.org/content/suppl/2016/04/26/128.11_suppl_1.S105.DC1

Permissions: Requests for permissions to reproduce figures, tables, or portions of articles originally published in _Circulation_ can be obtained via RightsLink, a service of the Copyright Clearance Center, not the Editorial Office. Once the online version of the published article for which permission is being requested is located, click Request Permissions in the middle column of the Web page under Services. Further information about this process is available in the Permissions and Rights Question and Answer document.

Reprints: Information about reprints can be found online at:
http://www.lww.com/reprints

Subscriptions: Information about subscribing to _Circulation_ is online at:
http://circ.ahajournals.org/subscriptions/
SUPPLEMENTAL MATERIAL

Detailed Methods

Patients and cell culture

Human left atrial appendages and blood samples were obtained from patients undergoing clinically-indicated heart surgery after informed consent. All protocols were approved by the University of Ottawa Heart Institution Research Ethics Board. Inclusion criteria for tissue donors selected patients between the ages of 18 and 80 who required cardiac surgery for coronary artery bypass grafting and/or valve surgery. Exclusion criteria included chronic infectious diseases (HIV, hepatitis), pregnant women or active sepsis.

CSCs were cultured as described previously. Cardiac tissue was minced, enzymatically digested with collagenase (1mg/mL; Invitrogen) and plated as cardiac explants on fibronectin coated dishes within cardiac explant media (CEM; Iscove’s Modified Dulbecco’s Medium (Invitrogen), 20% fetal bovine serum (FBS) (Invitrogen), 100 U/ml penicillin G, 100 ug/ml streptomycin (Invitrogen), 2 mmol/l L-glutamine (Invitrogen) and 0.1 mmol/l 2-mercaptoethanol (Invitrogen)). After seven days in culture, the heterogeneous population of cells (termed cardiac outgrowth) that spontaneously emigrated from the plated tissue was harvested using mild trypsinization (0.05% trypsin; Invitrogen). Three additional harvests were collected every 7-9 days. Only CSCs collected during the second harvest were used in this study.

CACs (or early outgrowth endothelial progenitor cells) were isolated from peripheral blood samples as described previously. Mononuclear cells were isolated using density-gradient centrifugation (Histopaque 1077; Sigma-Aldrich, Canada) and placed in culture for 4-6 days in endothelial basal media (EBM-2; Clonetics, Canada) supplemented with EGM-2-MV-SingleQuots (Clonetics) that included 5% FBS, 50 ng/ml human vascular endothelial growth factor and other necessary supplements.
factor (VEGF), 50 ng/ml human insulin-like growth factor-1 (IGF-1) and 50 ng/mL human epidermal growth factor (EGF). CACs were harvested by mechanical dissociation and were used for experimentation within seven days of culture.

**Conditioned media**

Conditioned media was obtained from CSCs, CACs and NHDFs after 48 hours of culture in hypoxic conditions (1% oxygen) to simulate the environment of the infarcted myocardium. Cells were seeded at 90% confluency (2.0 \times 10^5 CSCs, 3.0 \times 10^6 CACs and 2.0 \times 10^5 NHDFs) in low serum basal media (Iscove’s Modified Dulbecco’s Medium, 1% fetal bovine serum (FBS), 100 U/ml penicillin G, 100 \mu g/ml streptomycin, 2 mmol/l L-glutamine and 0.1 mmol/l 2-mercaptoethanol) on 6-well plates (Corning). To examine the relationship between CAC and CSC in co-culture conditions, conditioned media was collected from CSC/CAC co-cultures at different confluency ratios that corresponded to half the number of cells used in either single stem cell system (CSC\text{low}/CAC\text{high} 5.0 \times 10^4/1.5 \times 10^6; CSC\text{high}/CAC\text{low} 1.0 \times 10^5/7.5 \times 10^5; CSC\text{high}/CAC\text{high} 1.0 \times 10^5/1.5 \times 10^6). After 48 hours in culture, protein content of the cell lysate was determined using standard Bradford techniques (BioRad, Canada).

**Cytokine arrays and ELISAs**

Cytokine secretion in conditioned media was screened using a custom protein array (Human Cytokine Antibody Array G Series kit; RayBiotech, USA) according to the manufacturer’s directions. The relative fluorescent signal was analyzed using a Genepix 4000B scanner with proprietary software (Molecular Devices Inc., USA). Median fluorescent values were normalized to the internal positive and negative controls. To detect growth factors that were secreted in excess to the negative cellular control (NHDF), the grouped NHDF densitometry signal was subtracted from individual CACs and CSCs values. Levels of cytokines of interest were confirmed using commercially available enzyme-linked immunosorbent (ELISA)
assay kits (R&D Systems, USA; angiogenin (DAN00), epidermal growth factor (EGF; DEG00),
hepatocyte growth factor (HGF; DHG00), interleukin-6 (IL-6; D6050), stromal cell-derived factor-
1α (SDF-1α; DSA00) and vascular endothelial growth factor (VEGF; DVE00)). All
immunosorbent measures were normalized to the protein content and media volume.

**In vitro angiogenic differentiation and cell migration**

The capacity of CACs and CSCs to stimulate angiogenic growth was assessed using a
growth factor depleted matrigel assay (ECM625, Millipore) in accordance with the
manufacturer’s instructions, 2.0 x 10⁴ human umbilical vein endothelial cells (HUVECs; Lonza)
were seeded on matrigel with stem cell conditioned media (CSC-alone, CAC-alone, CSC/CAC
co-cultures), NHDF conditioned media (negative cellular control) or serum free DMEM (negative
media control). Random fields were analyzed using phase microscopy after 18 hours
incubation in normoxic culture at 10x magnification. Cumulative tubular growth was determined
using Image J software plug-in, NeuronJ (National Institutes of Health (NIH);
http://rsb.info.nih.gov/ij).¹

Similarly, the capacity of conditioned media to recruit CACs was assessed using
fibronectin coated trans-well plates (24 wells, 3.0 µm pores; Corning) with 3.0 x 10⁴ CACs
plated in the upper well in serum-free DMEM while conditioned media (CSC-alone, CAC-alone,
CSC/CAC co-cultures, NHDF) was placed in the bottom well. Serum free DMEM containing
100 ng VEGF was used as an unbiased control to normalize individual variations in CAC
migration. After 24 hours of normoxic incubation, the inserts and the remaining upper
compartment CACs were removed. CACs that had successfully migrated through the
polycarbonate membrane were fixed (4% paraformaldehyde) and stained with DAPI (Sigma-
Aldrich). Fluorescent microscopy (10x magnification, 6 random fields) was used to determine
the average number of cells per random field (ImageJ, ICTN plugin, Center for Bio-Image Informatics, USA).

**In vitro cardiac differentiation**

Cells were cultured for seven days in cardiogenic media (CGM; DMEM-LG, 40% MCDB-201, 0.75% dimethylsulfoxide, 0.1% 10 mmol/l L-ascorbic acid, 0.01% ITS liquid media supplement, 0.01% linoleic acid-albumin, 0.01% Pen-Strep, 0.0002% 0.25 mmol/l dexamethasone, 0.001% 2-mercaptoethanol, 10 ng/ml recombinant mouse fibroblast growth factor 8b, 100 ng/ml fibroblast growth factor 4, 10 ng/ml recombinant human protein rhDKK-1 and 10 ng/ml recombinant human bone morphogenetic protein 2). After seven days in culture, cells underwent immunohistochemistry or quantitative PCR screening for markers of cardiac identity. RNA was extracted (70022, Qiagen), treated with DNase and cDNA was synthesized (Roche). Gene-specific dual labeled qPCR primers and probes were designed for qPCR using a Roche LightCycler 480II thermocycler.

To verify protein expression, cells were stained with monoclonal antibodies specific to proteins expressed by terminally differentiated cells including α-smooth muscle actin (α-SMA; ab7817, Abcam), cardiac troponin T (cTnT; ab10214, Abcam) and Von Willebrand factor (vWF; ab8822, Abcam). Six random fields in each well from three separate cultures were examined and the mean fluorescence intensity for each lineage markers was normalized to the cell count (DAPI signal) for statistical comparison. Exposure time, fluorescent intensity and contrast settings were kept constant between all samples of a specific fluorochrome. Images were analyzed using image J software to measure the relative fluorescent intensity compared to the background signal. Intensity values were then normalized to DAPI signal intensity taken from the same random field to control for cell number in each field.

**Flow cytometry of transplanted cells**
The phenotypic profile of the CACs and CSCs transplanted into NOD SCID mice was confirmed using flow cytometry (FACSAria I, BD Biosciences, USA). Cells were fixed with 4% paraformaldehyde and stored at 4°C. Monoclonal antibodies and similarly conjugated isotype-matched control monoclonal antibodies for CD34 (316401, eBioscience), CD90 (555596, BD Biosciences), c-Kit (FAB332A, RD Systems), CD133 (130-090-826, Miltenyi) and VEGFR2 (FAB357P, RD Systems) were used. A minimum of 100,000 events were collected after performing fluorescent compensation using single labelled controls. Positive cells were defined as the percent of the population falling above the 99th percentile of the isotype control (Cyflogic, v.1.2.1 CyFlo Ltd, USA).

**Myocardial infarction, cell injection, and functional evaluation**

Myocardial infarctions (MI) were performed in male NOD-SCID mice (8-9 weeks old) by permanent ligation of the left anterior descending (LAD) coronary artery. Animals were injected with buprenorphine (0.05mg/kg; subcutaneous) one hour prior to surgery and twice daily thereafter for 3 days. During the ligation, mice were incubated and anesthetized using isoflurane (maintained at 2-3%) and upon closure, animals were injected with 0.5 cc of saline (subcutaneous).

Seven days after LAD ligation, 10-15 mice per group were injected with $1 \times 10^5$ cells (CSCs, CACs or NHDF) or a negative vehicle control (PBS) at the cardiac apex and lateral border zone. A fourth group received CAC and CSC co-administration at the cardiac apex and lateral border zone, with both CACs and CSCs ($0.5 \times 10^5$ CSCs + $0.5 \times 10^5$ CACs) as a single cell therapy. Trans-thoracic intra-myocardial injection was performed using echocardiographic guidance to confirm cells were injected into the myocardium. Twenty one days, 28 days and 16 weeks (long-term cohort only) after LAD ligation, the effect of cell therapy was evaluated from the left ventricular ejection fraction (LVEF; VisualSonics V1.3.8, VisualSonics, Toronto,
Canada). Animals were sedated using a ketamine/xylazine (100mg/ml / 20mg/mL) cocktail (10µL/g; intraperitoneal injection) during each ECHO procedure as well as during intramyocardial injections. To account for multiple comparisons made from the serial echocardiograms, this functional data was analyzed using a repeated measures mixed model with post-hoc testing done using t-tests, as appropriate, with Bonferonni’s correction.

**Histology**

After the final assessment of myocardial function, the mice were sacrificed. The hearts were excised, fixed with 4% paraformaldehyde, embedded in OCT and sectioned. Tissue viability within the infarct zone was calculated from Masson’s trichrome (Invitrogen, Canada) stained sections by tracing the infarct borders manually and then using ImageJ software to calculate the percent of viable myocardium within the overall infarcted area. To evaluate stem cell engraftment and differentiation, immunostaining for human nuclear antigen (HNA; SAB4500768, Sigma, Canada) was used to detect cells of human origin. Co-staining with non-specific α-SMA (ab125266; Abcam), cTnT (ab66133; Abcam) and vWF (11778-1-AP; Proteintech Group, USA) was used to identify cells that differentiated into functional cardiomyocytes. Capillary density within the infarct border zone was assessed by staining for non-specific isolectin B4 expression (B-1205; Vector Laboratories, Canada) in conjunction with DAPI (Sigma, Canada). The total number of nuclei within one image field of the border zone were counted and assessed for isolectin B4 expression.

**References**


Supplemental Figures and Figure Legends

**Supplement Figure S1.** CAC and CSC surface marker expression. Flow cytometry analysis of the relative proportion of surface marker expression on representative fractions of CSCs and CACs.
<table>
<thead>
<tr>
<th>A</th>
<th>B</th>
<th>C</th>
<th>D</th>
<th>E</th>
<th>F</th>
<th>G</th>
<th>H</th>
<th>I</th>
<th>J</th>
<th>K</th>
<th>L</th>
<th>M</th>
<th>N</th>
</tr>
</thead>
<tbody>
<tr>
<td>POS 1</td>
<td>POS 2</td>
<td>POS 3</td>
<td>NEG</td>
<td>NEG</td>
<td>Angiogenin</td>
<td>Angiopoietin-1</td>
<td>Angiostatin</td>
<td>AR</td>
<td>bFGF</td>
<td>BMP-4</td>
<td>BMP-6</td>
<td>BMP-7</td>
<td>EGF</td>
</tr>
<tr>
<td>POS 1</td>
<td>POS 2</td>
<td>POS 3</td>
<td>NEG</td>
<td>NEG</td>
<td>Angiogenin</td>
<td>Angiopoietin-1</td>
<td>Angiostatin</td>
<td>AR</td>
<td>bFGF</td>
<td>BMP-4</td>
<td>BMP-6</td>
<td>BMP-7</td>
<td>EGF</td>
</tr>
<tr>
<td>E-Selectin</td>
<td>FGF-4</td>
<td>FGF-6</td>
<td>FGF-7</td>
<td>FGF-9</td>
<td>GCSF</td>
<td>GM-CSF</td>
<td>HB-EGF</td>
<td>HGF</td>
<td>IFN-gamma</td>
<td>ICAM-1</td>
<td>IGFBP-1</td>
<td>IGFBP-2</td>
<td>IGFBP-3</td>
</tr>
<tr>
<td>E-Selectin</td>
<td>FGF-4</td>
<td>FGF-6</td>
<td>FGF-7</td>
<td>FGF-9</td>
<td>GCSF</td>
<td>GM-CSF</td>
<td>HB-EGF</td>
<td>HGF</td>
<td>IFN-gamma</td>
<td>ICAM-1</td>
<td>IGFBP-1</td>
<td>IGFBP-2</td>
<td>IGFBP-3</td>
</tr>
<tr>
<td>IGFBP-4</td>
<td>IGFBP-5</td>
<td>IGFBP-6</td>
<td>IGF-I</td>
<td>IGF-II</td>
<td>IL-1alpha</td>
<td>IL-1beta</td>
<td>IL-10</td>
<td>IL-6</td>
<td>LIF</td>
<td>L-Selectin</td>
<td>MCP-1</td>
<td>M-CSF</td>
<td>MIP-3alpha</td>
</tr>
<tr>
<td>IGFBP-4</td>
<td>IGFBP-5</td>
<td>IGFBP-6</td>
<td>IGF-I</td>
<td>IGF-II</td>
<td>IL-1alpha</td>
<td>IL-1beta</td>
<td>IL-10</td>
<td>IL-6</td>
<td>LIF</td>
<td>L-Selectin</td>
<td>MCP-1</td>
<td>M-CSF</td>
<td>MIP-3alpha</td>
</tr>
<tr>
<td>MMP-2</td>
<td>MMP-9</td>
<td>Osteopontin</td>
<td>Osteoprotegerin</td>
<td>PDGF-AA</td>
<td>PDGF-AB</td>
<td>PDGF-BB</td>
<td>PECAM-1</td>
<td>SCF</td>
<td>SDF-1alpha</td>
<td>SDF-1beta</td>
<td>TGF-alpha</td>
<td>TGF-beta 1</td>
<td>TGF-beta 2</td>
</tr>
<tr>
<td>MMP-2</td>
<td>MMP-9</td>
<td>Osteopontin</td>
<td>Osteoprotegerin</td>
<td>PDGF-AA</td>
<td>PDGF-AB</td>
<td>PDGF-BB</td>
<td>PECAM-1</td>
<td>SCF</td>
<td>SDF-1alpha</td>
<td>SDF-1beta</td>
<td>TGF-alpha</td>
<td>TGF-beta 1</td>
<td>TGF-beta 2</td>
</tr>
<tr>
<td>TGF-beta 3</td>
<td>TIMP-1</td>
<td>TIMP-2</td>
<td>TNF-alpha</td>
<td>TNF-beta</td>
<td>VCAM-1</td>
<td>VEGF</td>
<td>VEGF R2</td>
<td>Neg</td>
<td>Neg</td>
<td>Neg</td>
<td>Neg</td>
<td>Neg</td>
<td>POS 2</td>
</tr>
<tr>
<td>TGF-beta 3</td>
<td>TIMP-1</td>
<td>TIMP-2</td>
<td>TNF-alpha</td>
<td>TNF-beta</td>
<td>VCAM-1</td>
<td>VEGF</td>
<td>VEGF R2</td>
<td>Neg</td>
<td>Neg</td>
<td>Neg</td>
<td>Neg</td>
<td>Neg</td>
<td>POS 2</td>
</tr>
</tbody>
</table>

**Supplement Figure S2.** Schematic of the custom protein array.
Supplement Figure S3. Long term (16 week) effects of CAC and CSC transplantation upon myocardial function. A. Week 16 echocardiograms demonstrate the long term effects of CAC and CSC transplantation upon LVEF. B. Quantitive PCR for human alu sequences demonstrating the modest long term (16 week) engraftment of first generation CAC and CSC therapies. *p≤0.05 vs. pre-transplant (day 7) LVEF; †p≤0.05 vs. PBS treatment at week 16; **p≤0.05 vs. CAC transplantation.
Supplement Figure S4. Long term (16 week) effects of CAC and CSC transplantation on left ventricular scar burden. A. Representative Masson’s trichrome images of each cell therapy 16 weeks after myocardial infarction. B. Percentage scar formation within the left ventricle was assessed using ImageJ software. CAC/CSC co-transplantation resulted in decreased scar burden when compared to single cell therapy, while all three therapies demonstrated enhanced effects over NHDF treated animals. * p≤0.05 vs. single-cell therapies ** p≤0.05 vs. all other cell therapies.
Supplement Figure S5. Capillary density within the border zone of the ventricular infarcts 28 days after cell transplantation. A. Representative images of the capillary density between treatment groups one microscope field from the infarct border zone. Scale bar = 30 µm. B. Comparison of the percentage of isolectin B4 positive capillaries within the border zone between treatment groups. CAC/CSC co-transplantation resulted in increased capillary density when compared to single cell therapy, while all three therapies demonstrated enhanced effects over control animals. * p≤0.05 vs. single-cell therapies ** p≤0.05 vs. all other cell therapies.
Supplement Figure S6. Lineage fate of retained human stem cells 28 days after transplantation. Random field analysis from histological sections demonstrating the co-segregation of human nuclear antigen positive cells with markers of cardiac (cTnT), smooth muscle (αSMA) or endothelial (vWF) identity. * p≤0.05 vs. vWF⁺/HNA⁺ expression in CAC treated hearts. † p≤0.05 vs. αSMA⁺/HNA⁺ or cTnT⁺/HNA⁺ expression in CSC and CAC+CSC treated hearts.
### Supplemental Tables and Table Legends

<table>
<thead>
<tr>
<th></th>
<th>All patients (N=53)</th>
<th>In vitro studies (n=44)</th>
<th>In vivo studies (n=9)</th>
</tr>
</thead>
<tbody>
<tr>
<td>Age (yrs)</td>
<td>68±2</td>
<td>68±2</td>
<td>67±3</td>
</tr>
<tr>
<td>BMI (kg/m²)</td>
<td>29±1</td>
<td>29±2</td>
<td>30±2</td>
</tr>
<tr>
<td>Gender (%male)</td>
<td>69%</td>
<td>67%</td>
<td>78%</td>
</tr>
<tr>
<td>Diabetes</td>
<td>37%</td>
<td>40%</td>
<td>25%</td>
</tr>
<tr>
<td>Hypertension</td>
<td>74%</td>
<td>76%</td>
<td>63%</td>
</tr>
<tr>
<td>Dyslipidemia</td>
<td>65%</td>
<td>67%</td>
<td>56%</td>
</tr>
<tr>
<td>Ongoing Smoking</td>
<td>10%</td>
<td>12%</td>
<td>0%</td>
</tr>
<tr>
<td>Thyroid disease</td>
<td>9%</td>
<td>8%</td>
<td>14%</td>
</tr>
<tr>
<td>Peripheral vascular disease</td>
<td>10%</td>
<td>5%</td>
<td>38%</td>
</tr>
<tr>
<td>Coronary Artery Disease</td>
<td>75%</td>
<td>70%</td>
<td>88%</td>
</tr>
<tr>
<td>History of MI</td>
<td>22%</td>
<td>21%</td>
<td>25%</td>
</tr>
<tr>
<td>Valvular Heart Disease</td>
<td>31%</td>
<td>33%</td>
<td>14%</td>
</tr>
<tr>
<td>Congestive Heart Failure</td>
<td>31%</td>
<td>33%</td>
<td>14%</td>
</tr>
<tr>
<td>NYHA class</td>
<td>1.7±0.2</td>
<td>1.6±0.2</td>
<td>2.0±0.6</td>
</tr>
<tr>
<td>LV ejection fraction</td>
<td>44±13</td>
<td>43±13</td>
<td>53±4</td>
</tr>
<tr>
<td>CCS class</td>
<td>1.0±1.3</td>
<td>0.8±1.2</td>
<td>2.8±0.5*</td>
</tr>
<tr>
<td>Creatinine (umol/L) / GFR (mL/min)</td>
<td>93±4/0.9±0.1</td>
<td>96±4/0.9±0.1</td>
<td>78±7/1.2±0.1*</td>
</tr>
</tbody>
</table>

Medications:
- Anti-platelet therapy: 75% (in vitro), 73% (in vivo), 100% (in vivo)
- Beta-blocker: 69% (in vitro), 74% (in vivo), 45% (in vivo)
- Statins: 61% (in vitro), 62% (in vivo), 57% (in vivo)
- ACEI or ARB: 82% (in vitro), 86% (in vivo), 57% (in vivo)

**Supplement Table S1.** Baseline clinical characteristics of the patients. BMI = Body mass index. MI = Myocardial Infarction. NYHA = New York Heart Association. LV = Left Ventricle. CCS = Canadian Cardiovascular Society. GFR = Glomerular Filtration Rate. ACEI = Angiotensin-Converting Enzyme Inhibitors. ARB = Angiotensin Receptor Blockers. Significant: p ≤ 0.05 vs. *in vitro* study patient characteristics.
<table>
<thead>
<tr>
<th>Time post MI</th>
<th>Group</th>
<th>End Diastolic Volume (µL)</th>
<th>End Systolic Volume (µL)</th>
<th>Stroke Volume (µL)</th>
<th>Ejection Fraction (%)</th>
</tr>
</thead>
<tbody>
<tr>
<td>1 week</td>
<td>NHDF</td>
<td>69.8±5.6</td>
<td>48.5±3.9</td>
<td>22.3±2.2</td>
<td>31.8±1.2</td>
</tr>
<tr>
<td></td>
<td>PBS</td>
<td>60.4±4.3</td>
<td>41.7±3.3</td>
<td>18.7±1.3</td>
<td>31.2±1.2</td>
</tr>
<tr>
<td></td>
<td>CAC</td>
<td>64.7±2.8</td>
<td>46.2±2.3</td>
<td>18.6±1.2</td>
<td>29.1±1.5</td>
</tr>
<tr>
<td></td>
<td>CSC</td>
<td>63.6±3.8</td>
<td>45.1±2.6</td>
<td>18.6±1.7</td>
<td>29.9±1.2</td>
</tr>
<tr>
<td></td>
<td>CAC+CSC</td>
<td>58.5±3.2</td>
<td>40.1±2.1</td>
<td>18.34±1.8</td>
<td>28.7±1.2</td>
</tr>
<tr>
<td>3 weeks</td>
<td>NHDF</td>
<td>79.8±11.2</td>
<td>41.5±9.7</td>
<td>18.3±1.8</td>
<td>24.0±1.6</td>
</tr>
<tr>
<td></td>
<td>PBS</td>
<td>74.3±7.4</td>
<td>57.7±6.6</td>
<td>16.6±1.2</td>
<td>23.3±1.7</td>
</tr>
<tr>
<td></td>
<td>CAC</td>
<td>90.3±5.6</td>
<td>61.1±5.1</td>
<td>29.2±2.1*</td>
<td>33.7±2.1*</td>
</tr>
<tr>
<td></td>
<td>CSC</td>
<td>83.3±6.1</td>
<td>52.9±5.6</td>
<td>30.4±2.2*</td>
<td>35.7±2.1*</td>
</tr>
<tr>
<td></td>
<td>CAC+CSC</td>
<td>77.1±2.8</td>
<td>42.9±1.8</td>
<td>34.1±2.2*</td>
<td>41.8±1.5**</td>
</tr>
<tr>
<td>4 weeks</td>
<td>NHDF</td>
<td>85.6±10.0</td>
<td>66.4±8.7</td>
<td>19.1±1.8</td>
<td>23.1±1.3</td>
</tr>
<tr>
<td></td>
<td>PBS</td>
<td>81.4±7.7</td>
<td>63.5±6.5</td>
<td>18.0±1.5</td>
<td>22.7±1.4</td>
</tr>
<tr>
<td></td>
<td>CAC</td>
<td>88.4±5.4</td>
<td>57.7±4.8</td>
<td>30.8±1.4*</td>
<td>35.2±1.8*</td>
</tr>
<tr>
<td></td>
<td>CSC</td>
<td>87.7±7.8</td>
<td>57.1±5.4</td>
<td>30.6±3.5*</td>
<td>36.1±2.1*</td>
</tr>
<tr>
<td></td>
<td>CAC+CSC</td>
<td>75.4±5.1*†</td>
<td>43.5±2.8</td>
<td>31.9±2.7*</td>
<td>41.9±1.4**</td>
</tr>
<tr>
<td>16 weeks</td>
<td>NHDF</td>
<td>124.5±37.9</td>
<td>104.1±37.7</td>
<td>20.4±1.2</td>
<td>19.8±3.8</td>
</tr>
<tr>
<td></td>
<td>PBS</td>
<td>85.0±13.7</td>
<td>70.8±11.7</td>
<td>14.2±2.3</td>
<td>16.0±1.8</td>
</tr>
<tr>
<td></td>
<td>CAC</td>
<td>92.4±5.5</td>
<td>62.4±4.9</td>
<td>30.0±3.1*</td>
<td>32.2±2.3*</td>
</tr>
<tr>
<td></td>
<td>CSC</td>
<td>84.8±7.5</td>
<td>56.8±6.1</td>
<td>27.9±1.2*</td>
<td>33.5±1.5*</td>
</tr>
<tr>
<td></td>
<td>CAC+CSC</td>
<td>89.7±5.2</td>
<td>55.5±4.2</td>
<td>34.2±1.3*</td>
<td>38.7±1.6**</td>
</tr>
</tbody>
</table>

**Supplement Table S2.** Echocardiographic measurements of left ventricle over 16 week follow-up period. *p≤0.05 vs. PBS or NHDF treatment; †p≤0.05 vs. CAC and CSC treatment.